Historic, Archive Document

Do not assume content reflects current scientific knowledge, policies, or practices.

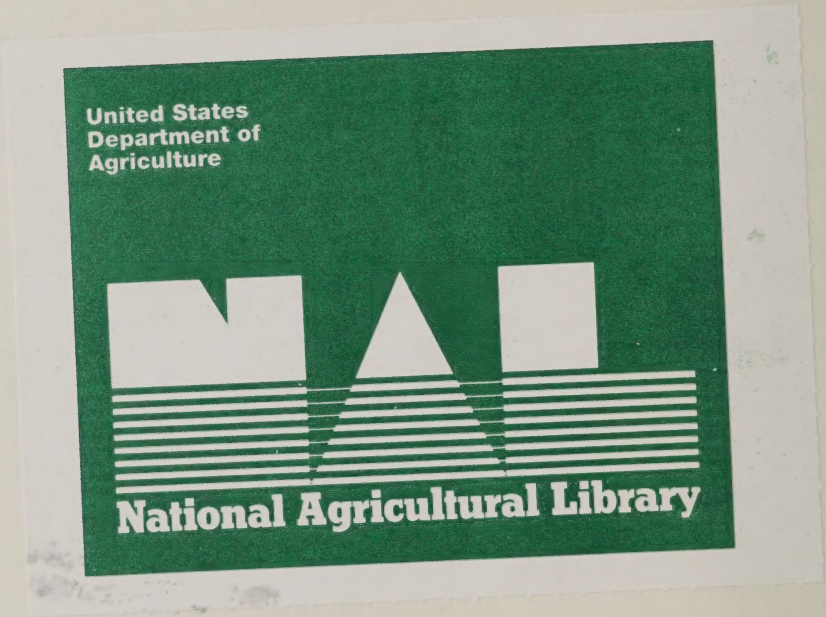
aSB933
.3
.S7

ed States
rtment of
culture

nce and
ation
inistration

Status of Biological Control of Filth Flies

A 106.2:F64


United States
Department of
Agriculture
National Agricultural Library

.3
.S7

STATUS OF BIOLOGICAL CONTROL OF FILTH FLIES

Proceedings of a Workshop

February 4-5, 1981
University of Florida
Gainesville

Sponsored by
Insects Affecting Man and Animals Research Laboratory,
Agricultural Research, Science and Education Administration,
U.S. Department of Agriculture

and

Institute of Food and Agricultural Sciences,
Cooperative Extension Services,
Department of Entomology and Nematology,
University of Florida

Organizing Committee

R. S. Patterson, Chairman
P. G. Koehler, P. B. Morgan, and R. L. Harris

U.S. DEPARTMENT OF AGRICULTURE
NATIONAL AGRICULTURAL LIBRARY
OCT 24 1996
CATALOGING PREP.

Science and Education Administration
U.S. Department of Agriculture
July 1981

This publication was reproduced from camera-ready copy supplied by the authors, who take responsibility for any errors in their papers. The use of trade names does not constitute a guarantee, warranty, or endorsement of products or services by the U.S. Department of Agriculture. The opinions expressed by the authors are not necessarily those of the U.S. Department of Agriculture.

This publication is available from the Insects Affecting Man and Animals Research Laboratory, P.O. Box 14565, Gainesville, Fla. 32604.

Published by Agricultural Research (Southern Region), Science and Education Administration, U.S. Department of Agriculture, P.O. Box 53326, New Orleans, La. 70153. Issued July 1981.

CONTENTS

FOREWORD

This workshop was initially planned following discussions with several research and extension scientists who were concerned about what to recommend to county agents and farmers about purchasing biocontrol organisms and traps for fly control. Some farmers had spent a considerable amount of money purchasing various organisms and devices with little or no improvement in their fly problem, while others raved about their purchases and claimed complete fly suppression. From a scientific point of view, we could not give endorsements based on testimonials, nor could we condemn a trap or organism without testing its merit. There was not enough scientific evidence to support most commercial field releases of parasites or use of traps. Therefore, we brought together the research, extension and commercial people to try to learn the state of development of fly biocontrol and the needs and problems of commercial production and distribution. This workshop was the first time that many researchers had a chance to meet and talk with the commercial people and listen to some of their problems, needs, and wants.

It was a difficult task to select speakers for this program, as many researchers have worked on various aspects of fly biocontrol. Because of the basic nature of the research, the organizing committee did not include any speakers on pathogens or the physiological aspects of host-parasite interrelationships. Our approach was more to the applied aspects of fly biocontrol in integrated pest management (IPM) schemes. Except for one, all the talks given at the workshop are presented here. We would like to note that all papers were reviewed by at least three reviewers, but that most papers have been reproduced directly from the material submitted by the authors, who are solely responsible for their contents.

As chairman of the workshop, I wish to thank all the speakers and participants who willingly gave of their time and money to attend. I also would like to thank the introductory speakers, Dean F. A. Wood, Dr. L. W. Larson, Dr. D. E. Weidhaas, and Dr. D. L. Shankland, for taking time from their busy schedules and starting the workshop off on the right foot. The workshop could never have been carried out or the proceedings completed without the help of Mrs. H. Amrhein, Mrs. J. Bickner, Mrs. B. Pritt, and Mrs. B. Hollien--all of whom receive special thanks. To

Dr. M. McGowan and Mr. K. Crosby, who assisted with setup of meetings and driving, and to the various technicians from the USDA Fly Biology and Control Unit and University of Florida Extension Service who also helped with the pickup and delivery of participants, thanks.

R. S. Patterson
Chairman, Organizing Committee

THE COORDINATION OF SCIENTIFIC AND COMMERCIAL ASPECTS OF BIOLOGICAL CONTROL OF FILTH BREEDING FLIES

J. J. Drea, Jr., and E. G. King

U.S. Department of Agriculture

Biological control is an established, yet continually developing, management strategy that is safe, efficient, and often very cost effective. The literature is voluminous, with examples of successful biological control projects, including the use of natural enemies to control filth breeding flies (Clausen 1978). The scientists at this Workshop here in Gainesville are the leaders in this area and certainly are to be congratulated.

It is impossible to define biological control in terms that are acceptable to all. Whatever your definition is, it certainly includes the concept of deliberate management of natural enemies as well as the importation and establishment of exotic parasites and predators. Included in this concept are the rearing and periodic release of natural enemies to control pests such as filth breeding flies.

Agricultural Research (AR), of the Science and Education Administration, supports research in biological control to develop technology that will hasten the successful intergration of biological control into a pest management system.

However, the effective integration of biological control into many factors that must be considered such as (1) the level of control that is required to be cost effective; (2) the type of natural enemies available for use; (3) the environmental condition into which the beneficial organisms are to be introduced; (4) the level of the technology that has been developed for the rearing, storage, shipment, and distribution of the natural enemy utilized; and (5) the type of other control measures being applied against the target pest or closely associated organisms.

Unfortunately for us, pest species are often hardy organisms that defy all our efforts to destroy them. Conversely, the

beneficial organisms are often extremely fragile and defy all our attempts to keep them alive. As a result, reliance upon the use of biological agents to control pests should be limited to those situations where scientifically and economically sound procedures can be utilized. This requirement is not unrealistic, but it does depend upon mutual cooperation between science and industry to arrive at a goal that is beneficial to the consumer.

Those of us who are researchers have the responsibility for developing scientifically sound procedures for the manipulation, utilization, and evaluation of the natural enemy. Those of us who are concerned with the commercial aspects of biological control must utilize the technology that has been developed by the scientists and by industry itself. The use of this technology must be realistic, recognizing that there are limitations which must be respected.

Any biological control project depends upon cooperation and coordination to be successful. These two factors are essential to the project from its inception to the final utilization by the general public.

Those of us who have worked overesas are well aware of our dependence upon others. We need support and coordination from our domestic counterparts or all our efforts overseas are useless. If there is a weak link in the chain that stretches from the foreign explorer through the domestic receiving station and eventually to the consumer in the U.S., the whole project falls apart, and the entire effort is lost. At the overseas location, there is a complex web of support and coordination that requires input from our own Government and that of the host Government. This may be the use of laboratory space and vehicles, or an exchange of information and specimens, or even working side-by-side in the field with another American or foreign scientist. This unified effort usually leads to the discovery and introduction into the U.S. of one or more beneficial organisms for use in our biological control efforts. However, the need for support and especially coordination does not end with the discovery of a new natural enemy.

Research concerned with the release and evaluation of a natural enemy in the field is very dependent upon cooperation between the scientists and the growers and private organizations in order to maximize the impact of the beneficial upon the target pest. Usually, this is not a problem. However, at times situations can arise in which the scientist, extension specialist, industry, and the consumer are not in agreement. This may arise through a lack of communication, or an over expectancy of results produced by the beneficials, or the results obtained are not immediately up to the level expected. Unfortunately, at times the results never attain the levels desired. Some of

this failure may be ascribed to the need for more time (biological control can be slow), the need for more beneficials, or more research, and a more realistic attitude on the part of those involved.

Some of these hurdles will be overcome through direct cooperation between scientists, extension, suppliers, and consumers. We wish to emphasize that recommendations are still the task of the extension specialists. Therefore, the contact between the scientist and the extension organization is especially important. We must listen to each other and decide what is the best way to produce results that are beneficial for all of us. If we do not, the grower, farmer, or rancher eventually will stop listening to all of us. If that happens, the project may be doomed.

We have worked both overseas and in the U.S. with Musca autumnalis De Geer, one of the filth breeding flies, but it was by no means extensive. We did succeed in establishing a staphylinid beetle predator, Aleochara tristis (Gravenhorst), in the U.S. This insect originated in France. At present, the impact of the beetle on the fly population in the U.S. is unclear. Unless colleagues present here can shed more light on its efficacy, it appears that the beetle is not very helpful. In any event, the USDA is now planning additional research overseas to obtain other dung-inhabiting insects in an attempt to control or, at least, reduce fly populations in the U.S. Like most research in biological control, it will be slow and will require several years for completion. One of our colleagues, Mr. H. Hoyer, from the European Parasite Laboratory, Sevres, France, will discuss this later in the program. In the meantime, we must continue our efforts to utilize those natural enemies we now have. To do so effectively will require additional research, coordination of our efforts, and continued contact between the scientists, extension servises, the commercial firms, and the renchers or growers. We have differences and will continue to do so, but we must work together to settle these differences. If we do not, the grower himself will settle them for us.

Hopefully, this Conference will (1) clarify the status of the technology for the use of natural enemies to control filth breeding flies and will (2) develop strategies for more fully developing and implementing this technology. By so doing, we all will benefit, and support for biological control will be available in the future for this and other projects. (A policy statement entitled "Status and Potential for Production and Use of Biological Agents for Pest Control" was developed by AR in 1978 to establish the position of AR on issues that arose with a Trichogramma project. The issues are similar to those addressed at this meeting. A number of copies of this report are available for distribution to those who may wish to have one.)

REFERENCE

Clausen, C. P. (ed.).
1978. Introduced parasites and predators of arthropod pests and weeds: a world review. USDA, Agric. Handb. 480; 545 pp.

IMPROVING COMMERCIAL BIOLOGICAL CONTROL OF FILTH FLIES WITH PARASITES

E. F. Legner

University of California, Riverside

That endophilous eusynanthropic flies (wholly dependent on humans for continuous survival) (Legner et al. 1974) have been lowered in density through periodic mass releases of parasitic insects is a scientific fact (Legner and Brydon 1966; Legner and Dietrick 1972; Morgan et al. 1975a, 1975b, 1976; Olton and Legner 1975; Rutz and Axtell 1979). This was further substantiated in reports by several speakers at this conference. The parasites most effective against endophilous flies are species of Spalangia and Muscidifurax, and, occasionally, Pachycrepoideus vindemiae Rondani and Tachinaephagus zealandicus Ashmead. Mass releases with 4 species have been stressed: Muscidifurax raptor Girault and Sanders, M. zaraptor Kogan and Legner, Spalangia endius Walker and Tachinaephagus zealandicus. However, to insure that mass releases are wholly effective, biotic and abiotic requirements such as humidity, temperature, habitat-depth and host preferences of these parasites must be considered (Ables 1975, Ables and Shepard 1976, Ables et al. 1976, Gerling and Legner 1968, Legner 1977, Legner and Olton 1971, Legner et al. 1976, Markwick 1974, Mourier 1971, Olton and Legner 1974, Tingle and Mitchell 1975).

Commercial insectaries recently have begun to sell parasites for fly control, often without regard to parasite behavioral differences, seasons of optimum activity, or even the correct identity of the parasites offered for sale. During the past 4 years, examination of material from 8 independent insectaries revealed that the Spalangia endius advertised for sale actually was primarily Nasonia vitripennis Walker, a common laboratory contaminant. This parasite, an exophilous eusynanthrope (Legner et al. 1974) seeks out habitats containing Calliphora, Phormia, Sacrophaga and Phaenicia spp. (see Legner 1967a), where the endophilous flies, Musca domestica L., Fannia canicularis L. and Stomoxys calcitrans (L.), usually do not breed. Thus, no logical basis exists to sell Nasonia vitripennis to control house and stable flies in

their principal breeding habitats. Indeed, Nasonia was once mass released by the millions in a gigantic biological control effort against 2 of its natural hosts, the sheep maggot fly (Phaenicia cuprina Wiedemann) in Australia and Calliphora stygia (F.) in New Zealand, that resulted in no significant fly reduction (Froggatt 1919; Froggatt and McCarty 1914; Miller 1922, 1927; Miller et al. 1936; Wilson 1960). Occasionally, Muscidifurax species are sold under the name of Spalangia endius or they comprise a significant proportion of the product labeled as the latter species.

Spalangia endius probably is the most capable of significantly reducting fly density (Morgan et al. 1976). Muscidifurax species caused only ca. 50% reduction when released during cool seasons (Legner and Brydon 1966, Legner and Dietrick 1972). Unfortunately, Spalangia species as a group, unlike Muscidifurax species and the contaminant Nasonia, are difficult to rear. Spalangia spp. require a higher humidity and temperature than the other species of fly parasites and are inferior in intrinsic competition with them within a puparium (Ables and Shepard 1974; Legner, unpub. data; Markwick 1974). In cases of multiple parasitism, Spalangia species invariably are destroyed by the other 2 parasites. Thus, a laboratory colony can quickly be displaced by either Muscidifurax or Nasonia, with the latter being competitively superior by virtue of its shorter developmental period and gregariousness. It must be emphasized that this superiority is shown only under laboratory culture conditions; in nature the habitats, searching capacity and penetration depth differ (Legner 1967b, 1977).

To improve the effectiveness of parasite releases, insectaries might seek periodic voluntary certification by federal or state authorities, as identification of the different parasite species demands considerable professional skill (Legner et al. 1976). For example, the Ricon-Vitova Insectaries of Ventura, California, have often provided samples from their cultures for examination by me. On several occasions, their parasite colonies where thus saved by detecting contamination. Simple culling procedures employed at the right time saved the integrity of the product and the reputation of the company, with minor costs in time and money.

The usefulness of a particular parasite species must be verified in different climates. Most of the scientific work with parasite mass releases has been conducted in the subtropics. In temperate latitudes there is a notable lack of scientific data to judge the potential effectiveness of parasite releases. Different parasite species apparently are more effective in different climates (Legner and Olton 1971).

Other more cryptic and little understood influences affect the performance of different parasite species. These influences

must be understood by commercial insectaries before their customers will be confident that the parasites they buy will lower fly densities. For example, in the suspected East African native range of Musca domestica and Stomoxys calcitrans, where breeding occurs in wild animal dung, Spalangia endius is the predominant parasite attacking endophilous flies in the dung of wild black buffalos (Syncerus caffer) and hippos (Hippopotamus amphibius) (Lugner, unpub. data). Spalangia cameroni Perkins and Spalangia nigroaenea Curtis predominate in accumulated dairy manure (Legner and Greathead 1969). Thus, the activity of certain parasites may be influenced by animal manure changed physiologically by diet. Other unknown causes also may help to explain this difference.

Parasite mass releases also may have no effect in situations where fly density is already extremely low. During a 7-month study on 6 California poultry ranches in 1980, 3 ranches that received biweekly releases of Spalangia endius were no different from 3 control ranches in the densities of Musca domestica, Fannia canicularis and Fannia femoralis Stein that they contained. These ranches practiced good manure management (Legner et al. 1973), had abundant predators present and what could be regarded as "scarce" fly densities (less than 1 fly caught per sticky fly tape per 24 h). All fly control programs should strive to guarantee the maximum abundance of natural predators. There is good evidence that high predation of eggs and young larvae may result in relatively low pupal population densities in which maximum parasitism has been observed (Legner 1971; Legner et al. 1975, 1980). Delicate interrelationships exist between parasitic insects, arthropod predators and their hosts when in balance at low densities (see Legner 1969).

Mass releases of parasites seem to be effective in well managed accumulations of animal wastes as found beneath caged poultry or in manure mounds on dairies. However, there have been no positive data to indicate that mass releases are effective in pig and calf pens, horse corrals, and feed lots, although these situations are productive breeding sources of flies. Parasite effectiveness against such species as Drosophila, biting gnats, horse flies, horn flies and face flies which also has been claimed by commercial insectaries, is unsupported by experimental evidence. New importations of predatory insects from Asia and Africa may ultimately prove most effective against some of the field dung breeding species (Legner 1978).

Much more experimental study thus is needed before greater reliance can be placed on parasite releases in a variety of habitats against a larger number of fly species. Clearly, more parasitic species should be tested and more careful attention should be given to different strains of parasite species (Legner 1977, Legner and Olton 1968). But first and foremost, the identity and quality of the parasite cultures being sold must be obtained and maintained at a high level.

REFERENCES

Ables, J. R.
1975. Bioecological relationships between hymenopterous parasitoids and their house fly host. Ph.D. Dissertation. Clemson University, Clemson, South Carolina. 48 pp.

Ables, J. R., and Shepard, M.
1974. Responses and competition of the parasitoids Spalangia endius and Muscidifurax raptor (Hymenoptera: Pteromalidae) at different densities of house fly pupae. Can. Entomol. 106(8): 825-830.
1976. Influence of temperature on oviposition by the parasites Spalangia endius and Muscidifurax raptor. Environ. Entomol. 5(3): 511-513.

Ables, J. R.; Shepard, M.; and Holman, J R.
1976. Development of parasitoids, Spalangia endius and Muscidifurax raptor in relation to constant and variable temperature: simulation and validation. Environ. Entomol. 5(2): 329-332.

Froggatt, J. L.
1919. An economic study of Nasonia brevicornis, a hymenopterous parasite of muscid Diptera. Bull. Entomol. Res. 9: 257-262.

Froggatt, W. W., and McCarthy, T.
1914. The parasite of the sheep-maggot fly (Nasonia vitripennis). Notes and observations in the field and laboratory. N.S.W. Agric. Gaz. 25(9): 759-764.

Gerling, D., and Legner, E. F.
1968. Developmental history and reproduction of Spalangia cameroni, parasite of synanthropic flies. Ann. Entomol. Soc. Am. 61(6): 1436-1443.

Legner, E. F.
1967a. The status of Nasonia vitripennis as a natural parasite of the house fly, Musca domestica. Can. Entomol. 99(3): 308-309.
1967b. Behavior changes the reproduction of Spalangia cameroni, S. endius, Muscidifurax raptor, and Nasonia vitripennis (Hymenoptera: Pteromalidae) at increasing fly host densities. Ann. Entomol. Soc. Am. 60(4): 819-826.
1969. Distribution pattern of hosts and parasitization by Spalangia drosophilae (Hymenoptera: Pteromalidae). Can. Entomol. 101(5): 551-557.
1971. Some effects of the ambient arthropod complex on the density and potential parasitization of muscoid Diptera in poultry wastes. J. Econ. Entomol. 64 (1): 111-115.
1977. Temperature, humidity and depth of habitat influencing host destruction and fecundity of muscoid fly parasites. Entomophaga 22(2): 199-206.
1978. Natural enemies imported in California for the

biological control of face fly, Musca autumnalis De Geer, and horn fly, Haematobia irritans (L.). Proc. Calif. Mosq. Vector Control Assoc., Inc. 46: 77-79.

Legner, E. F.; Bowen, W. R.; McKeen, W. D.; Rooney, W. F.; and Hobza, R. F.
1973. Inverse relationship between mass of breeding habitat and synanthropic fly emergence, and the measurement of population densities with sticky tapes in California inland valleys. Environ. Entomol. 2(2): 199-205.

Legner, E. F., and Brydon, H. W.
1966. Suppression of dung-inhabiting fly populations by pupal parasites. Ann. Entomol. Soc. Am. 59(4): 638-651.

Legner, E. F., and Dietrick, E. J.
1972. Inundation with parasitic insects to control filth breeding flies in California. Proc. Calif. Mosq. Control Assoc., Inc. 40: 129-130.

Legner, E. F., and Greathead, D. J.
1969. Parasitism of pupae in East African populations of Musca domestica and Stomoxys calcitrans. Ann. Entomol. Soc. Am. 62(1): 128-133.

Kegner, E. F.; Greathead, D. J.; and Moore, I.
1980. Population density fluctuations of predatory and scavenger arthropods in accumulating equatorial East African bovine excrement of three age classes. Bull Soc. Vector Ecol. 5: 23-44.

Legner, E. F.; Moore, I.; and Olton, G. S.
1976. Tabular keys and biological notes to common parasitoids of synanthropic Diptera breeding in accumulated animal wastes. Entomol. News 87(3,4): 113-144.

Legner, E. F., and Olton, G. S.
1968. Activity of parasites from Diptera: Musca domestica, Stomoxys calcitrans, and species of Fannia, Muscina, and Ophyra. II. At sites in the Eastern Hemisphere and Pacific Area. Ann. Entomol. Soc. Am. 61(5): 1306-1314.
1971. Distribution and relative abundance of dipterous pupae and their parasitoids in accumulations of domestic animal manure in the southwestern United States. Hilgardia 40(14): 505-535.

Ligner, E. F.; Olton, G. S.; Eastwood, R. E.; and Dietrick, E. J.
1975. Seasonal density, distribution and interactions of predatory and scavenger arthropods in accumulating poultry wastes in coastal and interior southern California. Entomophaga 20(3): 269-283.

Legner, E. F.; Sjogren, R. D.; and Hall, I. M.
1974. The biological control of medically important arthropods. Crit. Rev. Environ. Control 4(1): 85-113.

Markwick, N. P.
1974. A comparative study of four housefly parasites (Hymenoptera: Chalcidoidea). Ph.D. Thesis, Victoria Univ., Wellington, N. Zealand. 333 pp.

Miller, D.
1922. Sheep maggot fly control by insect parasites. The introduction of Nasonia brevicornis. N. Zealand J. Agric. 24: 211-212.
1927. Parasitic control of sheep maggot flies. Notes on current work. N. Zealand J. Agric. 34: 1-4.

Miller, D.; Clark, A. F.; and Dumbleton, L. J.
1936. Biological control of noxious insects and weeds in New Zealand. N. Zealand J. Sci. Tech. 18: 579-593.

Morgan, P. B.; Patterson, R. S.; and LaBrecque, G. C.
1976. Controlling house flies at a dairy installation by releasing a protelean parasitoid, Spalangia endius (Hymenoptera: Pteromalidae). J. Ga. Entomol. Soc. 11: 39-43.

Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Weidhass, D. F.; and Benton, A.
1975a. Suppression of a field population of houseflies with Spalangia endius. Science 189: 388-389.

Morgan, P. B.; Patterson R. S.; LaBrecque, G. C.; Weidhass, D. E.; Benton, A.; and Whitefield, T.
1975b. Rearing release of the house fly pupal parasite, Spalangia endius Walker. Environ. Entomol. 4(4): 609-611.

Mourier, H.
1971. Seasonal occurrence of pupal parasitoids of the housefly, Musca domestica (Diptera) in Denmark. Vidensk, Medd. Dan Naturchist. Foren. Ksh. 134: 109-118.

Olton, G. S., and Legner, E. F.
1974. Biology of Tachinaephagus zealandicus (Hymenoptera: Encyrtidae), parasitoid of synanthropic Diptera. Can. Entomol. 106(8): 785-800.

Rutz, D. A., and Axtell, R. C.
1979. Sustained releases of Muscidifurax raptor (Hymenoptera: Pteromalidae) for house fly (Musca domestica) control in two types of caged layer poultry houses. Environ. Entomol. 8(6): 1105-1110.

Tingle, F. C., and Mitchell, E. R.
1975. Seasonal reproduction of Muscidifurax raptor and Spalangia endius under simulated Florida conditions. Environ. Entomol. 4(3): 383-384.

Willson, F.
1960. A review of the biological control of insects and weeds in Australia and Australian New Guinea. Commonw. Inst. Biol. Control Tech. Commun. 1.

THE POTENTIAL USE OF PARASITES TO CONTROL *MUSCA DOMESTICA* L. AND OTHER FILTH BREEDING FLIES AT AGRICULTURAL INSTALLATIONS IN THE SOUTHERN UNITED STATES

Philip B. Morgan

U.S. Department of Agriculture

The common house fly (*Musca domestica* L.) is not a native of the Western Hemisphere. It probably originated in Africa and was transported to the Western Hemisphere during pre-Columbian times (Legner and McCoy 1966). Since Legner and Greathead (1969) observed the same species of parasites attacking fly puparium in Africa and in the Western Hemisphere, these parasites also probably had their origns in Africa but were transported throughout the world with the spread of the muscoid flies. However, parasites of Hymenoptera utilize the pre-imago stadia of house flies as a source of food. Many of these Hymenoptera are cosmopolitan and Legner and Greathead (1969) observed that some of these same Hymenopteran species parasitize fly pupae in east Africa.

The most effective species which attack fly pupae are in the family Pteromalidae. Some representatives that offer potential for fly control are: *Spalangia endius* Walker, *S. cameroni* Perkins, *S. nigra* Latreille, *S. nigroaenea* Curtis, *S. muscidarum* Richardson (=*S. nigroaenea*), *S. afra* Silvestri, *S. fallax* Masi, *S. gemina* sp. n., *S. longepetiolata* sp. n., *S. melanogastra* Masi, *S. obscura* sp. n., *S. seyrigi* Risbee, *S. simplex* Perkins, *S. sulcifera* sp. n., *Muscidifurax raptor* Girault and Sanders, *M. zaraptor* Kogan and Legner, *M. raptorellus* Kogan and Legner, *M. raptoroides* Kogan and Legner, *M. uniraptor* Kogan and Legner, *Nasonia vitripennis* (Walker), *Mormoniella vitripennis* (Walker) (=*N. vitripennis* (Walker)), *Pachycrepoideus vindemiae* (Rondani) (=*P. dubius* Ashmead) and *Tachineaphagus zealandicus* Ashmead, which is an effective larval parasite.

These insects that parasitize *M. domestica* were studied by Beard (1964), Legner (1967a, 1972), Legner et al. (1965, 1967), McCoy (1965), Legner and McCoy (1966), Legner and Gerling (1967), Legner and Olton (1968), Legner and Greathead (1969),

Ables and Shephard (1974a,b, 1976), Olton and Legner (1974), Morgan (1977), Morgan and Patterson (1975a,b), and Morgan et al. (1975a,b, 1976 a,b,c). Their use to control field populations of house flies was discussed by Legner and Brydon (1966), Legner and Dietrick (1972) and Keiding (1974). Those that parasitize *Stomoxys calcitrans* (L.) were reported by Pinkus (1913) and Monty (1972). Lindquist (1936), Combs and Haelscher (1969), Depner (1968) and Peck (1974) described parasites attacking *Haematobia irritans* (L.). The parasites of *Musca autumnalis* De Geer were studied by Turner et al. (1968), Burton and Turner (1968), Hayes and Turner (1971) and Hair and Turner (1965).

Female parasites are ready to mate and oviposit immediately upon emergence from the host puparia. Female wasps obtain their nourishment by ingesting the haemolymph of the host as it exudes from the oviposition site (Gerling and Legner 1968). The typical sequence of oviposition has 4 phases--finding the host area; locating host pupae; drumming and drilling; and feeding. For example, Edwards (1955) observed that once the female *N. vitripennis* found a pupae she systematically examines the surface while drumming with the tips of the antenna. Then she begins tapping with the tip of the abdomen on the surface of the puparium. This activity apparently places the tip of the ovipositor in position for drilling, a procedure that requires from 10 min. to 1 hr. When the wall of the puparium is pierced, the entire length of the ovipositor is inserted, and an egg is deposited on the developing pupa. Similar behavior has been noted with *S. endius, M. raptor* and *P. vindemiae*. All 3 species require from 10 to 15 min. to complete the cycle of host selection, drumming, drilling, oviposition and feeding. The *S. endius* and *M. raptor* apparently do not mark the oviposition site. However, the *P. vindemiae* female will touch the area around the oviposition site 5 or 6 times with the tip of the abdomen. Laboratory studies have also shown that *M. raptor* does not recognize oviposition sites of other *M. raptor* (Morgan 1980a) and *M. raptor* will parasitize host pupae previously parasitized by *S. endius*. The *M. raptor* larvae will outcompete all of the immature stadia of *S. endius*.

Apparently, there is variation in the number of eggs laid per pupae and the number that will develop. Wiley (1971a) observed that as many as 25 larvae of *N. vitripennis* can mature on 1 house fly pupae, while Whiting (1967) reported as many as 200 developing from 1 Sarcophaga pupa. Holmes (1972) found that genetically marked female *N. vitripennis* which were the second or third to oviposit on *S. huillata* Parker produced fewer progeny than marked females which were the first to oviposit on the host. Olton and Legner (1974) observed that 18 larvae of *T. zealandicus* can mature on 1 host. Laboratory studies have shown that both *M. raptor* and *S. endius* will make multiple ovipositions on 1 pupa. However, Morgan and Patterson (1975b) rarely observed more than 1 *M. raptor, S. endius* or *S. cameroni*

maturing on a host. Wiley (1971b) observed that *M. zaraptor* chose to oviposit on unparasitized *M. domestica* pupae rather than on pupae already parasitized by *N. vitripennis* or *S. cameroni*. He also noted that the discrimination was greater against parasitized pupae of *M. domestica* by *M. zaraptor* and least against those attacked by *S. cameroni*. After the parasite egg has been deposited, temperature has an effect on the developmental time of the parasite. For example, Gerling and Legner (1968) found the developmental time from initial oviposition by *S. cameroni* to eclosion of the adult parasite was 24-27 days at 26°C and 90-100% relative humidity (RH). Morgan et al. (1975a,b) documented a similar period for *S. endius*, and they found that by increasing the temperature to 27.8°C, the life cycle of *S. endius* was reduced to 20 days. However, they noted that 33-35 days are required for the life cycle of *S. endius* in the field. Additional laboratory studies have shown that *S. endius* development requires 14 days at 32.2°C, 19 days at 29.4°C, 21 days at 28.3°C, 24 days at 26.6°C, 25 days at 25.5°C, 29 days at 23.8°C and 34 days at 21.1°C at 70-80 RH Increasing the temperature above 32.2°C did not decrease the developmental time. Pinkus (1913) observed that *S. muscidarium* required a developmental time of 84 days at 13.9°C and on 2 occasions required 106 to 109 days. Legner and Gerling (1967) found that it required 19-22 days at 26°C and 90-100% RH. Morgan et al. (1978, 1979b) reported that it required a developmental time of 15 to 18 days and 19 to 21 days at 25.5°C and 60% RH for *P. vindemiae* and *M. raptor*, respectively. This concurs with Nstvik (1954) who concluded that developmental time for *P. vindemiae* was 19 days at 25°C and 65% RH and Crandall (1939) determined that 18 days was needed. Nagel and Pimental (1963) reported 14 days at 26.7°C for *N. vitripennis*. Whiting (1967) reported a generation time of 10 days at 28°C.

The average total progeny per female *N. vitripennis* was 139 (Nagel and Pimental 1963). Nstvik (1954) found the average total progeny per female *P. vindemiae* to be 298. Ables and Shephard (1974b) reported the average progenies per female *S. endius* and *M. raptor* were 15-40 and 5-30, respectively. Morgan et al. (1975a, 1976b) observed that 1 *S. endius* female produced an average of 9.46-9.6 progeny from 1- and 2-day-old house fly pupae and 16.1 progeny from 0- to 4-day-old stable fly pupae. They also reported 2.6 progeny per wasp per day and a male:female sex ratio of 1:2; unmated females produced males only. This correlated with an average lifespan of 3.88 days for each female *S. endius* when the wasps obtained their protein from the haemolymph of host pupae and with a 33.15% daily loss rate, which would allow an average of 36.7 ovipositions and 10 F_1 progeny per female *S. endius*. These data agree closely with those Lindquist (1936) obtained with *S. muscidarium* var. *stomoxysiae* Girault (=*S. endius*). Morgan et al. (1978, 1979b) observed that *M. raptor* females had a daily loss rate of 11%; the number of pupae parasitized and the number of progeny per female per day were 13 and 7.54,

respectively. The male:female ratio was 1:2.2 and the growth rate per generation was 55-fold. They also determined that the daily loss rate of *P. vindemiae* females was 52%. The number of pupae parasitized and the number of F_1 progeny per female per day was 5.7 and 2.7, respectively. The male:female ratio was 1:4.2, and the growth rate per generation was 2.1. Girault and Sanders (1910) reported sex ratios in *N. vitripennis* ranging from 1 male to 3 females and 1 male to 9 females. Nagel and Pimental (1963) observed that the longevity of *N. vitripennis* males averaged 1.62 days and that of the females averaged 6.96 days. The average progeny per female parasite was 139.8. Both Wylie (1967) and Legner (1969) concluded that total oviposition was correlated with high host densities and host size. When *S. endius* was exposed to *M. domestica* pupae at host:parasite ratios of 0.5:1 through 25:1, the percentage of parasitism decreased and the number of parasitized pupae increased as the ratio of pupae:parasites increased. There was a significant difference in the number of pupae parasitized per adult wasp and the number of progeny produced per adult wasp associated with the ratio of host:parasite. There was a significant difference in the number of pupae parasitized per adult wasp associated with age of pupae at ratios above 1:5, i.e. more for 1- and 2-day-old than 3-day-old pupae. When *S. endius* was offered a choice of pupae of *M. domestica* or *S. calcitrans* the percentage of parasitism was similar to that obtained when *M. domestica* or *S. calcitrans* pupae were exposed to the wasps (Morgan et al. 1979a).

In evaluating the activity of parasites against *M. domestica*, *S. calcitrans*, *Fannia canicularis* (L.) and *F. femoralis* (Stein) at sites in the Western Hemisphere, Legner (1967b) found that 92% of the house fly pupae collected in Uruguay were parasitized by 6 species of parasites (*M. raptor*, *S. cameroni*, *S. endius*, *S. nigroaenea*, *Tachinaephagus giraulti* Johnson and Tiegs and *Trichopria* n. sp.); in New Brunswick, Canada, 90.7% of the house fly pupae were parasitized by *M. raptor* and *S. nigroaenea*. He also found that a host such as *S. calcitrans* with the habit of pupating nearer the surface in the breeding site was generally more heavily parasitized and that the parasites did not disperse rapidly from a release site. In fact, dispersal was usually slow beyond host-free barrier zones, as from 1 poultry ranch to another.

In other tests made over a period of 18 months, Legner and Brydon (1966) evaluated parasitism at 2 poultry ranches in southern California. Although 6 species of parasites were active, *M. raptor* and *S. endius* accounted for 95% of observed parasitism in *F. femoralis* and *Ophyra leucostoma* (Wiedemann). Similarly, from March through June, 1970, Legner and Dietrick (1972) released thousands of *S. endius*, *M. raptor* and *T. zealandicus* at 6 poultry ranches to control *F. canicularis* and *F. femoralis*. Samples taken in June 1970 revealed a 6.5 times lower density (13.8 to 2.1) of these flies and the percentage

parasitism had almost doubled (12.9 to 22.5%). Additional inoculative releases of *S. endius*, *M. raptor*, *M. zaraptor* Kogan and Legner and *T. zealandicus* were made by Legner and Dietrick (1974) over a 20-month period on poultry ranches located in southern California. They observed significant reduction in average densities of Diptera: *M. domestica*, *Muscina stabulans* (Fallen), *F. canicularis*, *F. femoralis*, *O. leucostoma*, *S. calcitrans*, and *phaenicia*. Also, releases made during the spring had greater effect on fly populations than did similar releases in the summer. Olton and Legner (1975) made inoculative releases of *T. zealandicus*, *S. endius* and *M. raptor* from December through April in an enclosed poultry house in southern California. The result was 46% parasitism of *M. domestica* but only 16% of *F. femoralis*.

In addition, McCoy (1965) released *M. raptor* but found that parasitism of fly pupae never exceeded 25%. Mourier (1972) also released *S. cameroni* and *M. raptor* on 6 farms (10,000 parasites/farm) in northern Denmark. Although the parasite population built up faster than normal, it was still insufficient to reduce host populations to an acceptable level.

Monty (1972) released *S. endius*, *S. nigra*, *M. raptor*, *P. vindemiae* and *Sphegigaster* sp. against populations of muscoid flies on Mauritius. *Spalangia* sp. was recovered in greater numbers than any other species released: 68% of the house flies were parasitized and 44.4% of *S. calcitrans*. However, no parasites were recovered from *Stomoxys nigra* Macquart. The inundative releases decreased the populations of *S. calcitrans* and *M. domestica*, but the percentage of parasitism dropped as soon as the releases were stopped. He therefore concluded that the parasites, even when they are well established, cannot maintain themselves at densities high enough to effect control.

Morgan and Patterson (1977) found that when augmentative releases of *S. endius* were made for 13 weeks against a mixed population of filth breeding flies at a feeding station located in a 44-acre pasture and a 700-acre dairy farm in west Florida, that within 10 weeks all of the collected *M. domestica* pupae were parasitized. Moreover, by the 12th week all *S. calcitrans* pupae and after 13 weeks all *Physiphora aenea* (F.) pupae collected were parasitized. When another feeding station was established, 137 m away during the 6th week of releases, all of the *M. domestica* pupae collected from within and at the base of the trough were parasitized by the 13th week. During this same time interval parasitism of *S. calcitrans* pupae from the base of the trough ranged from 99-100%, and within the trough ranged from 33-88%. At a small commercial dairy in North Florida, Morgan et al. (1976c) released *S. endius* 3 times a week for 5 weeks, a total of 47,700 female parasites. By the end of the release all of the house fly pupae collected were parasitized and the fly population was reduced 93%.

In the test at a poultry installation containing 6,700 caged layers, Morgan et al. (1975a) released 44,520 *S. endius* females per week for 10 weeks. After 4 weeks, 100% parasitism was observed and the house flies were completely suppressed within 35 days. When *S. endius* was released at a beef farm over a period of 2-1/2 months for the control of *M. domestica* and *S. calcitrans*, they reduced the fly population 84%. Although 100% parasitism was obtained during this same time interval when similar releases were made at a swine farm, the *M. domestica* reduction was limited to 37% due to fly immigration from adjacent agricultural installations (Morgan 1980b).

It was observed by Morgan et al. (1981a) that when augmentative releases of *M. raptor* and *S. endius* were conducted at a 30,000 caged layer installation in North Florida, that *M. raptor* did not seek *M. domestica* pupae beneath the surface of the soil and did not function well in dry hot weather. *Spalangia endius* will, under similar conditions, successfully parasitize field pupae both above and below the surface of the soil but will be prevented from locating and effectively parasitizing pupae if the poultry droppings are not kept dry.

Although very successful results were obtained from the field releases conducted by Morgan and coworkers, the efficient use of augmentative releases of *S. endius* depends upon regulating the number of female parasites released in proportion to the absolute number of fly pupae in the population to be controlled. Initially, Weidhaas et al. (1977) using all available laboratory data and field data of *M. domestica* and *S. endius* were able to relate this information through a preliminary simulation model to the control developed by Morgan et al. (1975a,b; 1976c). In 1977, 1978 and 1979 Morgan et al. (1981a,b) conducted augmentative releases of *S. endius* at a 30,000 caged layer installation in North Florida. During this 3-year field study life history parameters were developed for the *M. domestica* population. The duration of the fly generation averages 15 days and consists of ca. 1 day for the egg stage; 1, 2 and 3 days for the 1-, 2- and 3-instar larvae, respectively; 4 days for the pupal stages and a 3-day preoviposition period, with oviposition on the 4th day. The adults life span averages 30 days and the total number of eggs laid by each female is ca. 1080, with 120 eggs per gonotrophic cycle or 40 eggs per ovipositing female per day. From this information a life table was constructed using the following formula and variables as given by Weidhaas and Haile (1978).

$$R_o = \frac{(\bar{S}_i{}^1)\ m\ (S_a{}^d)}{1 - (S_a{}^c)}$$

Where:

R_o = the growth rate per generation
S_a = the average daily survival of adult females

$\bar{S}_i$ = the average daily survival of immature stages
$\bar{l}$ = the development time in days
m = female eggs oviposited per day
d = the preoviposition period
c = oviposition cycle in days

In North Florida LaBrecque et al. (1972) estimated the growth rates of *M. domestica* ranged from 1x to 6x with an average of <3x. In table 1 (Morgan et al. 1981a) values of the parameters that produce growth ratios of 1x, 2.5x and 5x for a population of house flies are illustrated. Table 2 (Morgan et al. 1981a) illustrates the life table that was constructed using the parameters from table 1. Although arbitrary numbers are illustrated in table 2 for the 3 generations growth rates, they serve as a practical guideline for field releases. The total fly population was determined by collecting samples of 3rd-instar larvae and translating these numbers into 1-day-old pupae and newly emerged females flies via the life table. Releasing the female *S. endius* against an estimated number of 1- and 2-day-old pupae enabled a close prediction of the percentage parasitism that would be obtained.

In 1978 the mass culturing technique produced emerging *S. endius* females Monday through Thursday, and with a 33% daily loss rate it was never possible to maintain a high wasp population. As a result it took 8 weeks to obtain 99% parasitism. With some areas of the manure turned liquid by inundative rains, 100% parasitism was not achieved since the female wasps would not seek and parasitize fly pupae in these damp areas (Morgan et al. 1981a).

A modified mass culturing technique for *S. endius* was used in the 1979 release. Female parasites emerged daily each week. This enabled a high wasp population to be maintained. It took only 4 weeks to achieve 100% parasitism, basing the number of released *S. endius* on the estimated 1- and 2-day-old *M. domestica* pupal population (Morgan et al. 1981b). Also, in 1979 Morgan et al. (1981b) revised the technique for evaluating field parasitism. Through 1978, field pupae had been held in the laboratory for 10 days to allow the non-parasitized flies to emerge and to allow the parasites to develop beyond the egg stage. Although the method was reliable, it always involved a 10-day lag in the estimate of parasitism in the field. Therefore, in 1979 the following criteria were used when field pupae were evaluated for parasitism:

1. Presence of parasite eggs, larvae, pupae or non-emerged adults
2. Presence of an oviposition wound on the puparium
3. Haematomia-like wound on the body surface of the host pupae
4. The host pupae tissue has a jaundiced flaccid look

Table 1.--Values of the parameters that produce growth rates of 1x, 2.5x, and 5x for a population of house flies (λ = the daily growth rate)

R_o	$\bar{S}_i$	l	m	S_a	d	c	g	S_a/λ	S_i/λ
1.0	.7135457486	11	20	0.8	4	1	1.0	--	--
2.5	.7755292588	11	20	0.8	4	1	1.0505821	.7614826104	.7381900556
5.0	.8258705	11	20	0.8	4	1	1.0919291	.7326483001	.756432379

Table 2.--Number of *Spalangia endius* Walker in different developmental stages and daily age groups for 3 growth rates

Stage	Age in days	1X		2.5X		5X	
		No. per daily age class	No. per stage or instar	No. per daily age class	No. per stage or instar	No. per daily age class	No. per stage or instar
Eggs	1	1000	1000	1000	1000	1000	1000
Larvae							
1st instar	1	714	714	738	738	756	756
2nd instar	2	509	872	545	947	572	1004
	3	363	2162	402	2363	432	2522
3rd instar	4	259	576	297	678	327	762
	5	185		219		248	
	6	132		162		187	
Pupae	1	94	243	119	320	142	391
	2	67		88		107	
	3	48		65		81	
	4	34		48		61	
Adult females	0	24	73	35	99	46	123
	1	20		27		34	
	2	16		21		25	
	3	13		16		18	
Ovipositing females	4-30	50	50	50	50	50	50

Pupae positive for any of the above characteristics were scored as parasitized.

Thus, it has been demonstrated by Morgan and coworkers that augmentative releases of *S. endius* against known populations of *M. domestica* can produce essentially complete parasitism of 1- and 2-day-old pupae and suppression of the adult flies. It is quite probable that had the earlier researchers been releasing parasites against known populations of house flies, they would have obtained similar results. Although the majority of the parasite research conducted by Morgan and coworkers has been confined to evaluating *S. endius* against *M. domestica* and *S. calcitrans*, other species of filth breeding flies may occasionally be present. As shown by Morgan and Patterson (1977) pupae of a non-economical filth breeding fly, *P. aenea*, were present. The levels of parasitism demonstrated that the *P. aenea* were diluting some of the parasites away from the target *M. domestica* and *S. calcitrans* pupae. Therefore, it will be important to obtain additional data about pupal parasites other than *S. endius* and to study the integration of the augmentation procedures with other methods of control, and to determine the requirements for control when several species of muscoid flies breed in proximity.

REFERENCES

Ables, J. R., and Shephard, M.
1974a. Hymenopterous parasitoids associated with poultry manure. Environ. Entomol. 3: 884-886.
1974b. Responses and competition of the parasitoids Spalangia endius and Muscidifurax raptor (Hymenoptera: Pteromalidae) at different densities of house fly pupae. Can. Entomol. 106: 825-830.
1976. Seasonal abundance and activity of indigenous Hymenopterous parasitoids attacking the house fly (Diptera: Muscidae). Can. Entomol. 108: 841-844.

Beard, R. L.
1964. Parasites of muscoid flies. Bull. WHO 31: 491-493.

Burton, R. P., and Turner, E. C., Jr.
1968. Laboratory propagation of Muscidifurax raptor on face fly pupae. J. Econ. Entomol. 61: 1380-1383.

Combs, R. L., Jr., and Hoelscher, C. E.
1969. Hymenopterous pupal parasitoids found associated with the horn fly in northeast Mississippi. J. Econ. Entomol. 62: 1234-1235.

Crandall, H. A.
1939. The biology of Pachycrepoideus dubius Ashmead (Hymenoptera), a pteromalid parasite of Piophila casei Linne. Ann. Entomol. Soc. Am. 32: 632-654.

Depner, K. R.
1968. Hymenopterous parasites of the horn fly, Haematobia irritans (Diptera: Muscidae) in Alberta. Can. Entomol. 100: 1057-1060.
Edwards, R. L.
1955. The host-finding and oviposition behavior of Mormoniella vitripennis (Walker) (Hymenoptera: Pteromalidae), a parasite of muscoid flies. Behavior 7: 88-112.
Gerling, D., and Legner, E. F.
1968. Developmental history and reproduction of Spalangia cameroni, parasite of synanthropic flies. Ann. Entomol. Soc. Am. 61: 1436-1443.
Girault, A. A., and Sanders, G. E.
1910. The chalcidoid parasites of the common house or typhoid fly (Musca domestica L.) and its allies. Psyche 17: 9-28.
Hair, J. A., and Turner, E. C., Jr.
1965. Attempted propagation of Nasonia vitripennis on the face fly. J. Econ. Entomol. 58: 159-160.
Hayes, C. G., and Turner, E. C., Jr.
1971. Field and laboratory evaluation of parasitism of the face fly in Virginia. J. Econ. Entomol. 64: 443-448.
Holmes, H. B.
1972. Genetic evidence for fewer progeny and a higher percent males when Nasonia vitripennis oviposits in previously parasitized hosts. Entomophaga 17: 79-88.
Keiding, J.
1974. House flies (Musca domestica). In R. Pal and R. H. Wharton (ed.), Control of Arthropods of Medical and Veterinary Importance. 138 p. Plenum Press, New York and London.
LaBrecque, G. C.; Meifert, D. W.; and Weidhaas, D. E.
1972. Dynamics of house fly and stable fly populations. Fla. Entomol. 55: 101-106.
Legner, E. F.
1967a. The status of Nasonia vitripennis as a natural parasite of the house fly, Musca domestica. Can. Entomol. 99: 308-309.
1967b. Behavior changes the reproduction of Spalangia cameroni, S. endius, Muscidifurax raptor, and Nasonia vitripennis (Hymenoptera: Pteromalidae) at increasing fly host densities. Ann. Entomol. Soc. Am. 60: 819-826.
1969. Adult emergence interval and reproduction in parasitic Hymenoptera influenced by host size and density. Ann. Entomol. Soc. Am. 62: 220-226.
1972. Observations on hybridization and heterosis in parasitoids of synanthropic flies. Ann. Entomol. Soc. Am. 65: 254-263.

Legner, E. F.; Bay, E. D.; and McCoy, C. W.
1965. Parasitic natural regulatory agents attacking Musca domestica L. in Puerto Rico. J. Agric. Univ. PR 49: 368-376.

Legner, E. F.; Bay, E. C.; and White, E. B.
1967. Activity of parasites from Diptera: Musca domestica, Stomoxys calcitrans, Fannia canicularis, and F. femoralis, at sites in the western hemisphere. Ann. Entomol. Soc. Am. 60: 462-468.

Legner, E. F., and Brydon, H. W.
1966. Suppression of dung-inhabiting fly populations of pupal parasites. Ann. Entomol. Soc. Am. 59: 638-651.

Legner, E. F., and Dietrick, E. I.
1972. Inundation with parasitic insects to control filth breeding flies in California. Proc. 40th Annu. Conf. Calif. Mosq. Contr. Assoc., Inc. January 31-February 2. p. 129-130.
1974. Effectiveness of supervised control practices in lowering population densities of synanthropic flies on poultry ranches. Entomophaga 19: 467-478.

Legner, E. F., and Gerling, D.
1967. Host feeding and oviposition on Musca domestica by Spalangia cameroni, Nasonia vitripennis, and Muscidifurax raptor (Hymenoptera: Pteromalidae) influences their longevity and fecundity. Ann. Entomol. Soc. Am. 60: 678-691.

Legner, E. F., and Greathead, D. J.
1969. Parasitism of pupae in east African populations of Musca domestica and Stomoxys calcitrans. Ann. Entomol. Soc. Am. 62: 128-133.

Legner, E. F., and McCoy, C. W.
1966. The house fly, Musca domestica Linneaus, an an exotic species on the western hemisphere incites biological control studies. Can. Entomol. 98: 243-248.

Legner, E. F., and Olton, G. S.
1968. Activity of parasites from Diptera: Musca domestica, Stomoxys calcitrans, and species of Fannia, Muscina, and Ophyra. Ann. Entomol. Soc. Am. 61: 1306-1341.

Lindquist, A. W.
1936. Parasites of horn fly and other flies breeding in dung. J. Econ. Entomol. 29: 1154-1158.

McCoy, C. W.
1965. Biological control studies of Musca domestica and Fannia sp. on southern California poultry ranches. Proc. 33rd Ann. Conf. Calif. Mosq. Contr. Assoc., Inc. January. p. 40-42.

Monty, J.
1972. A review of the stable fly problem in Mauritius. Rev. Agric. Sucriere Maurice 51: 13-29.

Morgan, P. B.
1977. The parasitic wasp--A research update. Proc. 36th Ann. Fla. Poultry Institute, May. p. 10.
1980a. Mass culturing three species of microhymenopteran pupal parasites, Spalangia endius Walker, Muscidifurax raptor Girault and Sanders, and Pachycrepoideus vindemiae (Rondani) (Hymenoptera: Pteromalidae). VIII Renunion Nacional de Control Biologico. Tecomon, Colima, Mexico. April 22-25, 1980.
1980b. Sustained releases of Spalangia endius (Hymenoptera: Pteromalidae) for the control of Musca domestica L. and Stomoxys calcitrans (L.) (Diptera: Muscidae). J. Kans. Entomol. Soc. 53: 367-372.
Morgan, P. B.; Benton, A.; and Patterson, R. S.
1976a. The potential use of parasites to control flies in the Caribbean area. Virgin Islands Agric. and Food Fair. p. 43.
Morgan, P. B.; LaBrecque, G. C.; Weidhaas, D. E.; and Patterson, R. S.
1979a. Interrelationship between two species of muscoid flies and the pupal parasite Spalangia endius (Hymenoptera: Pteromalidae). J. Med. Entomol. 16: 331-334.
Morgan, P. B., and Patterson, R. S.
1975a. Field parasitization of house flies by natural populations of Pachycrepoideus vindemiae (Rondani), Muscidifurax raptor Girault and Sanders, and Spalangia nigroaenea Curtis. Fla. Entomol. 58: 202.
1975b. Possibilities of controlling stable flies and other muscoid flies with parasitic wasps. Proc. 46th Ann. Mtg. Fla. Anti-Mosq. Assoc., April 13-16. p. 29-35.
1977. Sustained releases of Spalangia endius to parasitize field populations of three species of filth breeding flies. J. Econ. Entomol. 70: 450-452.
Morgan, P. B.; Patterson, R. S.; and LaBrecque, G. C.
1976b. Host-parasitoid relationship of the house fly, Musca domestica L., and the protelean parasitoid, Spalangia endius Walker. J. Kans. Entomol. Soc. 49: 483-488.
1976c. Controlling house flies at a dairy installation by releasing a protelean parasitoid Spalangia endius Walker. J. Ga. Entomol. Soc. 11: 39-43.
Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Weidhaas, D. E.; and Benton, A.
1975a. Suppression of a field population of house flies with Spalangia endius. Science 189: 388-389.
Morgan, Philip B.; Patterson, R. S.; LaBrecque, G. C.; Weidhaas, D. E.; Benton, A.; and Whitfield, T.
1975b. Rearing and release of the house fly pupal parasite

Spalangia endius Walker. Environ. Entomol. 4: 609-611.

Morgan, P. B.; Weidhaas, D. E.; and LaBrecque, G. C.
1978. Host-parasite relationship of the house fly, Musca domestica L. and the microhymenopteran parasite, Pachycrepoideus vindemiae (Rondani). Southwestern Entomol. 3: 176-181.
1979b. Host-parasite relationship of the house fly, Musca domestica L., and the microhymenopteran pupal parasite, Muscidifurax raptor Girault and Sanders (Diptera: Muscidae and Hymenoptera: Pteromalidae). J. Kans. Entomol. Soc. 52: 276-281.

Morgan, P. B.; Weidhaas, D. E.; and Patterson, R. S.
1981a. Host-parasite relationship: Programmed releases of Spalangia endius Walker and Muscidifurax raptor Girault and Sanders, against estimated populations of Musca domestica L. J. Med. Entomol. 18: 158-166.
1981b. Host-parasite relationship: Augmentative releases of Spalangia endius Walker used in conjunction with population modeling to suppress field populations of Musca domestica L. J. Kans. Entomol. Soc. (In Press).

Mourier, H.
1972. Release of native pupal parasitoids of house flies on Danish farms. Vidensk. Medd. Dan. Naturhist. Foren. 135: 129-137.

Nagel, W. P., and Pimental, D.
1963. Some ecological attributes of a Pteromalid parasite and its house fly host. Can. Entomol. 95: 208-213.

Nstvik, E.
1954. Biological studies of Pachycrepoideus dubius Ashmead (Chalcidoidae: Pteromalidae), a pupal parasite of various Diptera. Oikos 5: 196-204.

Olton, G. S., and Legner, E. F.
1974. Biology of Tachinaephagus zealandicus (Hymenoptera: Encytridae) parasitoid of synantrophic Diptera. Can. Entomol. 106: 785-800.
1975. Winter inoculative releases of parasitoids to reduce house flies in poultry manure. J. Econ. Entomol. 68: 35-38.

Peck, O.
1974. Chalcidoid (Hymenoptera) parasites of the horn fly, Haematobia irritans (Diptera: Muscidae), in Alberta and elsewhere in Canada. Can. Entomol. 106: 473-477.

Pinkus, H.
1913. The life history and habits of Spalangia muscidarum Richardson, a parasite of the stable fly. Psyche 20: 148-158.

Turner, E. C., Jr.; Burton, R. P.; and Gerhardt, R. R.
1968. Natural parasitism of dung-breeding Diptera: a comparison between native hosts and an introduced host, the face fly. J. Econ. Entomol. 61: 1012-1015.

Weidhaas, D. E., and Haile, D. G.
1978. A theoretical model to determine the degree of trapping required for insect population control. Bull. Entomol. Soc. Am. 24: 18-20.

Weidhaas, D. E.; Haile, D. G.; Morgan, P. B.; and LaBrecque, G. C.
1977. A model to simulate control of house flies with a pupal parasite, Spalangia endius. Environ. Entomol. 6: 489-500.

Whiting, A. R.
1967. The biology of the parasitic wasp Mormoniella vitripennis (=Nasonia brevicornis) (Walker). Q. Rev. Biol. 42: 333-406.

Wylie, H. G.
1967. Some effects of host size on Nasonia vitripennis and Muscidifurax raptor (Hymenoptera: Pteromalidae). Can. Entomol. 99: 742-748.
1971a. Observations on intraspecific larval competition in three hymenopterous parasites of fly puparia. Can. Entomol. 103: 137-142.
1971b. Oviposition restraint of Muscidifurax zaraptor (Hymenoptera: Pteromaldae) on parasitized house fly pupae. Can. Entomol. 103: 1537-1544.

USE OF PREDATORS AND PARASITES IN FILTH FLY IPM PROGRAMS IN POULTRY HOUSING

Richard C. Axtell

North Carolina State University

INTRODUCTION

Poultry production is increasingly based on high density, confined-animal systems causing increasing problems of managing pest populations. In particular, the high bird densities result in potentially high fly populations accompanying the rapid accumulation of manure which is an excellent fly breeding medium. Typically, for each pound of feed consummed, a laying hen will deposit about an equal weight of wet droppings. This will amount to about a hundred pounds of wet droppings per year per laying hen (equal to an average of five pounds of wet droppings per dozen eggs). It is apparent that fly management depends upon manure management as an essential ingredient in an Integrated Pest Management (IPM) program for poultry production systems (Axtell 1970ab, 1981) Biological control agents (parasites and predators) along with selective use of insecticides are the other components in such an IPM program. The interrelationships between manure management, biological control agents and chemical control agents are important and strongly affected by the type of poultry housing and production systems. Understanding these relationships and the principles of fly management are essential to an understanding of the potential role of certain biological control agents. The biological control agents are not panaceas for the fly problem. Biological control agents should properly be viewed as components of a total fly management program.

POULTRY HOUSING AND MANURE MANAGEMENT

Poultry housing may be classified into three categories (caged-layer, breeder and broiler), in the case of chickens, in order of the severity of the fly problem. By far the greatest populations of flies occur in the caged-layer houses which are widely used for commercial egg production. There are generally

three (sometimes four) birds per cage (about 1.2 ft^2) with the cages stacked in two to four tiers on both sides of a walkway. This arrangement may be found in basically three types of caged-layer houses: narrow, wide-span and high-rise.

The narrow house has a single center walkway with two tiers of bird cages on each side, movable curtain side walls, and a dirt floor (occasionally concrete) upon which the manure accumulates under the cages. The narrow house is usually 10 ft wide and 300-400 ft long with several side-by-side at one farm. Each house contains about 5,000 birds. In the narrow house there is close contact between the accumulated manure and the outside environment. Manure is removed at infrequent intervals by simple methods such as by shovel, tractor-mounted scraper or front-end loader.

The wide-span caged-layer house has usually three or four walkways with two-tiered (occasionally three-tiered) cages. The sides may be movable curtains or enclosed with fans for ventilation control (an "environmental-control" house). Under the cages the manure accumulates on concrete floors (rarely dirt) which may be level or may be shaped into shallow pits between the walkways (i.e. under the cages). The manure under the cages may be removed infrequently by tractor-mounted scraper or may be removed at regular intervals (once or twice a week) by means of a scraper mounted on tractor or pulled by a built-in cable system or by flushing (often daily) with large volumes of water from a built-in piping system. Ironically, the flush systems when properly designed and managed, essentially eliminate the fly problem, but often create a problem of mosquitoes breeding in the waste lagoon (Rutz and Axtell 1978, Rutz, Axtell and Edwards 1980). The wide-span houses in which the manure is allowed to accumulate are very much like the narrow houses except that the center rows of manure are in less contact with the external environment. Those houses which have the manure removed frequently (daily or 2-3 times a week) attempt to provide fly control by elimination of the breeding medium. Often this is successful, but sometimes the manure removal systems break down or fail to remove all of the manure and fly breeding becomes a serious problem. The wide-span houses are typically 30-40 ft wide, 300-400 ft long and contain 12,000-18,000 birds.

The high-rise type of caged-layer house is a two-story structure with the top floor the typical cage arrangement of a wide-span house, i.e. three to five walkways with tiered cages on each side. The area beneath the cages is open so that the droppings fall onto the ground of the first floor. This arrangement allows accumulation of manure for several years, if desired. A tractor-mounted front-end loader is used to remove the manure. The first floor is usually at ground level, but occasionally (especially in northern areas) may be set part way

into the ground. High-rise houses may have open sides with movable curtains or closed sides with ventilating fans. The high-rise house is sometimes called a deep-pit house. Since this house is designed for long term manure accumulation (2-4 years) fly management depends upon proper manure management through several fly seasons. These houses are generally 30-40 ft wide, 300-400 ft long and housing about 30,000 birds.

The second most prolific fly breeding occurs in the breeder houses. Most of these are broiler-breeder houses because of the need for continuous production of large number of broilers for processing. Typically the broiler-breeder houses are wide-span (30 ft wide X 350 ft long) with the birds (about 7,000) running free. The center one-third of the house has litter (wood shavings) on the dirt floor and is unsuitable for production due to the dryness. The outer one-third along each side has a slatted floor elevated 2-3 ft above the ground level. The feeders and waterers are on the slatted portions and most of the droppings accumulate beneath the slatted floor. Fly breeding occurs in that area of manure accumulation. The open sides with movable curtains allow for air exchange and limited contact between the manure and the external environment. About once every 10 months the flock is changed and the manure and litter removed from the house.

The third category of housing is the broiler houses (turkey houses are very similar). These houses are wide-span with the entire dirt floor covered with litter (wood shavings) and the birds free running. No fly breeding of significance occurs in these houses due primarily to the dryness. The litter is removed (at least partially) each time the broiler flock is changed (about every 4 months).

FLIES ASSOCIATED WITH POULTRY MANURE

The house fly, Musca domestica L. (Family Muscidae), is the major pest fly species associated with poultry manure. Management programs aimed at suppressing the numbers of house flies will usually concurrently suppress the number of other fly species which, in an improperly managed system, may become abundant enough to be pests. The house fly deposits batches of eggs in the areas of the poultry manure having the most attractive odors and moisture levels. One female fly can lay as many as six egg batches with 75-200 eggs per batch. The eggs hatch into the first-instar larvae in 12-24 hours and subsequently molt into a second- and third-instar. Larval development is completed in 4-7 days followed by the pupal stage which typically lasts 3-4 days. The pupal stage is within a dark brown, hardened and thickened case (called the puparium) which is formed from the integument of the third-instar larva. The life cycle (egg to adult) requires 10 days at 29°C, 21 days at 21°C and 45 days at 15°C. Adult flies typically live about 3-4 weeks, but can live as long as 8 weeks.

A related muscoid fly which is frequently found in large numbers in poultry houses is the black garbage fly, Ophyra leucostoma (Wiedemann). The life cycle is essentially the same as that of the house fly. The larva of Ophyra develops readily in poultry manure and, given the opportunity, will prey on other fly larvae. However, the adult Ophyra are pestiferous and the simple fact of their larvae being capable of predation on other fly larvae does not qualify them as a practical biological control agent.

Flies of the muscoid genus Fannia are common in poultry manure and often pestiferous although usually not to the extent of the house fly. F. canicularis (L.) is the most common, but F. femoralis (Stein) may be common in some houses. All have a flattened, brownish larvae with lateral and caudal spines. All have a life cycle similar to the house fly, but a slightly longer developmental time.

Soldier flies, Hermetia illucens (L.) (Family Stratiomyidae), deposit their eggs in poultry manure and the larvae, which are large and robust, may become extremely abundant. When large numbers of larvae are present, their activity and digestion of the manure sometimes renders the manure semi-liquid and physically unsuitable for survival of house fly larvae (Axtell and Edwards 1970). The larvae do not actually prey upon the house fly larvae as is sometimes erroneously claimed. The soldier fly larvae, even though they may reduce the numbers of house flies, are a serious pest because their liquifying of the manure makes the usual manure-removal procedures extremely difficult to accomplish. Further they apparently reduce the plant fertilizer qualities of the manure although this has not been quantified. The life cycle of the soldier fly is much longer than that of the house fly, being 40-75 days. The adult soldier fly is a large, wasplike, dark fly which is not very active and seldom is abundant enough to be a pest. Although soldier fly larvae on occasions become abundant and a problem in all types of caged-layer houses and under the slatted portions of broiler-breeder houses, they seem to be particularly abundant in the high-rise houses (at least in North Carolina) which defeats the original purpose of the high-rise, i.e. to allow accumulation and drying of the manure for several seasons.

A variety of species of blow flies (Family Calliphoridae) occur in poultry manure, but usually not in large enough numbers to constitute a major problem. Broken eggs and dead chickens in the manure are particularly attractive to the ovipositing females and are important factors in cases where substantial numbers of blow flies occur. The life cycles of blow flies are similar to the house fly although sometimes a few days shorter for larval development.

FLY PREDATORS ASSOCIATED WITH POULTRY MANURE

Accumulations of poultry manure which has the proper moisture and physical conditions contain large numbers of mites and beetles which prey on fly eggs and/or larvae. When the manure accumulates in a coned pile beneath caged-layers a gradient of moisture levels in the manure develops through the natural drying process. The various species of predators concentrate in the areas optimal for each species development. Together these species exert a large suppressive effect on the house fly population (and other flies).

Predaceous Mites

Three families of mites known to prey upon fly eggs and/or larvae naturally occur in poultry manure (Axtell 1961, 1963a). They are Macrochelidae, Uropodidae and Parasitidae. Typically the order of invasion in the spring is Parasitidae-Macrochelidae-Uropodidae. As the fly season progresses, mite numbers increase drastically. Overall, the order of abundance is Uropodidae-Macrochelidae-Parasitidae in poultry manure.

Macrochelidae. The most common machochelid mite in poultry manure is Macrocheles muscaedomesticae (Scopoli). This species, like most (but not all) macrochelids, has arrhenotokous reproduction, i.e. offspring from unfertilized eggs are all haploid males while those from fertilized eggs are diploid females. This provides a mechanism for maximizing species survival in precarious habitats. The macrochelid eggs hatch in 6-10 hours (27°C), producing a six-legged larval stage which molts producing the eight-legged protonymph in 6-11 hours. A second molt 13-24 hrs later, produces the deutonymph with a duration of 17-26 hours. A final molt produces the adult; thus 51-60 hours are required for development from egg to adult. Adult females have an average life span of 24 days. Depending upon substrate and food, a female will produce up to 25 eggs per day, but 4 to 5 is probably typical.

M. muscaedomesticae adult females are phoretic on flies which provides a mechanism for dispersal from old areas of manure to newer areas more likely to be breeding flies (Axtell 1964, Farish and Axtell 1971). As manure dries, its attractiveness to the mites decreases while concurrently the attractiveness of flies remains constant. Once the attractiveness of the manure is less than that of a visiting fly, the mite will grasp the fly and be carried off. Upon the fly visiting an area of fresh moist manure, the mite will detach due to the attractiveness of the fresh manure being greater than the attractiveness of the fly. Sensilla on the tarsi of the first pair of legs are the principle olfactory receptors (Farish and Axtell 1965, Coons and Axtell 1973). Attachment of the mite to the fly is by the chelicerae, but no feeding occurs. Stages of the mite other than the adult females are not phoretic.

Fly eggs and first-instar larvae are fed upon by M. muscaedomesticae protonymphs, deutonymphs and adults. Alternative foods are nematodes and acarid mites, both very common in manure. The adult mites prefer the fly eggs to nematodes while the reverse is true for the nymphal stage. The conditions will determine the numbers of fly eggs and/or larvae destroyed with up to about 20 eggs destroyed per day per mite demonstrated in some experiments. Substantial reductions in fly numbers in cow manure and poultry manure have been demonstrated under simulated field conditions. It has been well documented that large numbers of M. muscaedomesticae occur in properly managed manure and clearly are an important factor in the natural suppression of muscoid fly populations, especially the house fly (Axtell 1963b). A review of the literature and an assessment of Macrochelidae as biological control agents has been published (Axtell 1969).

In addition to M. muscaedomesticae, other species of macrochelids with very similar biology and predation behavior are found in animal manure (Axtell 1961, 1963b). Some similar species may be easily misidentified. Among the species, M. glaber (Müller) and Glyptholaspis confusa (Foà) are common fly predators in the United States while others, such as M. insignitus (Berlese) and M. peniculatus Berlese are common in Europe.

Uropodidae. The most commonly encountered uropodid mite predaceous on flies in manure is Fuscuropoda vegetans (DeGeer). F. vegetans is predaceous on first-instar fly larvae, being unable to subdue the larger and more vigorous second- and third-instar fly larvae (O'Donnell and Axtell 1965, Willis and Axtell 1968). This mite is usually unable to feed upon the fly egg. It feeds on manure-inhabiting nematodes as well as organic matter. F. vegetans aggregates in the manure and is slower moving than the macrochelid. The uropodid population increases slowly due to the 30-40 day life cycle, but often becomes the most abundant mite after 8-12 wks of manure accumulation. F. vegetans is phoretic (in a specialized deutonymphal stage) mainly on dung beetles and thus is dispersed to new areas of manure.

F. vegetans and M. muscaedomesticae, due to their behavior and distribution in the manure, are complimentary predators of flies (Willis and Axtell 1968). M. muscaedomesticae confines itself to the outermost layer of manure in areas likely to be used for egg deposition. The macrochelid prefers the egg stage, moves rapidly and has well developed olfactory sense. Those fly eggs which escape predation by the macrochelid hatch into a first-instar larvae which, being negatively phototropic, move deeper into the manure where the uropodid mites aggregate. The group attacks and gregarious feeding behavior of the uropodid tend to contain the fly larvae. F. vegetans does not destroy as many flies per individual as the macrochelid and the actual number destroyed would be strongly influenced by chance,

availability of alternate foods and the physical consistency of the manure.

Parasitidae. Very little is known about the parasitids associated with fly breeding in domestic animal manure. A species (Parasitus coleoptratorum L.) associated with dung beetles has been studied, but it apparently is not common in the manure of livestock production systems. A related undescribed species in the genus Poecilochirus has been found commonly in poultry and dairy cattle manure in confined quarters and its adults and deutonymphs are predators of fly eggs and first-instar larvae with preference for the larvae (Wise and Axtell 1969, Wise 1970). Further taxonomic study of the Parasitidae in manure is needed to determine whether we are dealing with primarily a single species or several.

The parasitids in manure appear early in the fly season and their populations decline later in the season. Thus they are apparently of short term importance as fly predators at the beginning of the fly season. Like the macrochelids and uropodids, the parasitids are phoretic on flies and beetles, but in the parasitids only the normal deutonymph stage is phoretic. The stimulus of fresh manure is required for normal development and molting to the adult stage. In old manure, the mite will remain in the deutonymph stage for long periods of time. The parasitid is fast moving and probably destroys many fly eggs and larvae, but we know too little about these mites to adequately assess them as biological control agents for filth flies.

Predaceous Beetles

Predaceous beetles, especially Histeridae and Staphylinidae, are common in poultry manure and are important consumers of fly eggs and larvae. The major species is the histerid, Carcinops pumulio (Erichson), which becomes very abundant and feeds readily on the immature stages of M. domestica and Fannia (Legner 1971, Peck 1969, Peck and Anderson 1969). It may be considered comparable in importance to the mite M. muscaedomesticae. The beetle fauna of poultry manure includes many other species, most of which are primarily scavengers, but many of which may occasionally feed upon the immature stages of flies. In North Carolina (Pfeiffer and Axtell 1980), C. pumulio, Alphitobius diaperinus (Panzer), Gnathoncus nanus (Scriba) and the Aleocharinae (Staphylinidae) are the most abundant. The diversity of beetles in narrow caged-layer houses tends to be greater than in high-rise houses. This is apparently due to the greater drying and heterogeneity (in moisture and texture) of the manure in the narrow houses. Although data are limited on predator rates, certain beetles (especially C. pumulio) should be considered important predators aiding in the suppression of fly production in poultry manure.

FLY PARASITES ASSOCIATED WITH POULTRY MANURE

Species Composition

Several species of hymenopterous parasites in the family Pteromalidae are found emerging from fly pupae in poultry manure (Rutz and Axtell 1980a). These are Muscidifurax raptor Girault and Sanders, Pachycrepoideus vindemiae (Rondani), Spalangia endius Walker, S. nigra Latreille, S. cameroni Perkins, S. nigroaenea Curtis, and Nasonia vitripennis (Walker). The N. vitripennis is a multiple parasite, i.e. many parasites will develop and emerge from a single fly pupae. The other are usually solitary parasites, i.e. a single parasite will develop and emerge from one fly pupae. In either case, the fly is killed by the parasite. N. vitripennis has been only sporadically recovered in our North Carolina studies and this species is generally considered a poor candidate as a biological agent for fly control under field conditions (Legner 1967).

In addition to the listed species, there are probably some others occurring in association with poultry manure, but not recognized due to the taxonomic difficulties in this group (Bouček 1963, Kogan and Legner 1970, Legner, Moore and Olton 1976). For example, in North Carolina we have occasionally collected a species (apparently undescribed) which is similar to, but not the same as, S. drosophilae Ashmead. Great care must be used to avoid incorrect identifications of specimens of Spalangia. It is particularly difficult to separate S. nigra and S. nigroaenea.

Relative Abundance

In an extensive survey of poultry houses in North Carolina, the most abundant of the fly parasites (in order) was M. raptor, S. cameroni, S. nigroaenea and S. endius. There were differences in relative parasite abundance among the Spalangia in the three climatic regions of the state (Mountains, Piedmont and Coastal Plain), but M. raptor was the most abundant in all of the regions. M. raptor was recovered throughout the year while Spalangia were collected from June through November. Overall, M. raptor was more abundant than the three most prevalent species of Spalangia combined (S. cameroni, S. nigroaenea and S. endius).

At newly established caged-layer poultry houses (Rutz and Axtell 1980b), there was invasion of several parasites within eight weeks, with M. raptor first followed by (in order): S. cameroni, P. vindemiae, N. vitripennis, S. endius and S. nigroaenea. Both M. raptor and P. vindemiae were prevalent during June through October, but were not abundant in July. S. cameroni was abundant in late summer and fall while S. endius and S. nigroaenea were most abundant in August. N. vitripennis was recovered in a few samples in June and July only.

The fly parasite fauna seems to be essentially the same at the different types of caged-layer and broiler-breeder poultry houses in North Carolina (Rutz and Axtell 1980a, 1981). Differences when they occur are due more to the degree of manure dryness and the related management practices rather than the type of house per se. In North Carolina it is most common for the parasite population to consist of mostly (up to 90%) M. raptor and S. cameroni. Proportions of these and other species may change with differences in the manure management. For example, in broiler-breeder houses the manure under the slatted portion of the house tends to be relatively dry with flies tending to pupate at greater depth than in the generally wetter manure of caged-layer houses (especially the high-rise type). Muscidifurax is claimed to be less active than Spalangia in searching deep in the manure, although further substantiation is needed (Ables and Shepard 1974, Legner 1977). Consequently, with fly pupae at greater depths, the Spalangia would be favored while with the fly pupae nearer the manure surface the M. raptor would be favored. Given equal opportunities to parasitize fly pupae, M. raptor is competively superior to Spalangia and will become the dominant species. This is apparent in laboratory colonies in which Spalangia are easily replaced by accidentally introduced M. raptor.

Parasitism Rates

The degree of fly parasitism resulting from these pteromalid parasites in poultry operations is difficult to evaluate. Three methods have been used: (1) recovery of naturally-occurring fly pupae, (2) placing emergence traps over the manure and (3) exposing sentinel fly pupae in mesh bags inserted into the manure. The finding of naturally-occurring pupae can be time-consuming and difficult with, as a result, data based often only on a few pupae. Further the age of the pupae (i.e. how long ago were they parasitized?) is not known. Emergence traps suffer from the same problems, i.e. it is guess work on where to place the trap, often very few parasites are recovered, and the age or number of pupae under the trap are not known. Further the trap may, by changing the conditions beneath it, tend to cause artificial parasitism rates by fly larvae and/or parasites moving under trap, but we have no data one way or the other on this factor. The pupal bag has the disadvantage of being artificial in the sense that the bag must be inserted into the manure. However, it has the tremendous advantage of being a standardized (by pupal age and numbers) sampling method. In my opinion, it is the best method available at this time.

In our various survey and parasite release experiments in North Carolina about 20-40% parasitism of sentinal fly pupae often has been found. The degree of parasitism varies greatly with season as well as the abundance and species of parasites present at a particular farm. Attempts to increase parasitism

by releases of M. raptor in North Carolina have been successful in two types of caged-layer houses and in broiler-breeder houses (Rutz and Axtell 1979, 1981). Concurrently with these releases, some degree of reduction in the numbers of house flies was shown in broiler-breeder houses and narrow caged-layer houses, but was not shown in a high-rise house. Similar release attempts in narrow caged-layer houses with an imported strain of S. endius did not result in an increase in parasitism by that species nor a decrease in the number of flies (Rutz and Axtell 1980b). The fact that the M. raptor strain was from North Carolina while the S. endius strain was from Florida suggested that house fly parasites need to be climatically adapted to the area where releases are to be made. Another factor also may be the fact that the S. endius strain had been in culture for several years while the M. raptor was in culture for only 1 or 2 years. It should be noted that the Florida S. endius strain was successfully used in a release program in Florida to drastically reduce fly numbers at a poultry farm (Morgan et al. 1975). In that Florida experiment the numbers released were very high (about 30 per bird per week) while in the North Carolina experiment, release was about 10 per bird per week. The successful releases with M. raptor in North Carolina were about 2 per bird per week at the caged-layer operations and about 5 per bird per week at the broiler-breeder operation.

ENHANCEMENT OF PREDATOR AND PARASITE POPULATIONS

Encouragement of the maximum heterogenity of naturally-occurring predators and parasites in poultry manure is the first step in the use of biological control agents for fly control (Anderson 1965, Axtell 1969). The key to this is manure management, i.e. maintaining the manure in the driest condition possible. This provides a desirable habitat for the predators to reproduce and to locate and attack fly eggs and larvae, it provides a habitat in which the parasites can locate and oviposite in the fly pupae, and it reduces the suitability of the manure for fly oviposition and larval development. Differences in the types of poultry housing affect the degree of manure drying and thus the degree of predator and parasite activity as well as the amount of fly breeding. The major factors are the amount and duration of manure accumulation and the amount of ventilation (air movement).

Manure Management

The amount of manure moisture is affected by four factors: (1) Leaking waterers.-Leaks in the poultry watering system is the major source of excess moisture in the manure; (2) Airflow. -The amount of air circulation should be kept high enough to dry the manure. This may be accomplished by using fans, open-sided houses, cutting vegetation which might impair air flow, and building the house on an elevated site having some air movement; (3) Drainage.-Runoff of rain water into a house can be

avoided by proper site selection, grading and drainage; (4) Substrate.-Underlying soil type affects the percolation and absorption of moisture from the manure in those types of housing lacking a concrete floor. In sandy soils it is often easier to promote manure drying than in areas with a heavy clay soil.

Populations of predators and parasites are affected by the manure removal schedule. Obviously in housing designed for complete removal of the manure every few days there is no build-up of the beneficial agents. In other types of housing, however, manure is allowed to accumulate for weeks or months (and even years in a properly managed high-rise house). The longer the manure can be allowed to accumulate and be kept relatively dry, the greater the opportunity for maximizing the effects of naturally occurring predators and parasites. When it becomes necessary to remove the manure after such long-term accumulations, only a part of the manure should be removed. A thick base of old manure should be left to perpetuate the predators and parasites as well as to assist in absorbing excess moisture from the newly added droppings. Rather than rapidly clean out a house at one time, it would be better to partially clean over a period of a few weeks. Such manure removal should be done in the cooler months when fly oviposition is not occurring. To be avoided is complete removal of the manure during the season of fly activity because the result will be a tremendous increase in numbers of flies 2-3 weeks after cleaning due to the destruction of the predator and parasite population, as well as the elimination of an absorptive base of old manure resulting in large areas of wet manure attractive for fly oviposition and larval development.

Selective Use of Insecticides

Unfortunately, insecticides which are effective against flies are generally about equally toxic to predators and parasites (Axtell 1966, 1968, Axtell and Edwards 1970). The slight differences in toxicities for some which can be demonstrated in the laboratory are not practical differences under field conditions. A possible exception is the compound N-cyclopropyl-1, 3,5-triazine-2,4,6-triamine (CGA 72662, Ciba-Geigy Corp) which is an effective fly control agent as either a feed additive or a larvicide spray, but appears to be relatively non-toxic to manure-inhabiting mites and beetles (Axtell, unpublished data).

Unless an insecticide is known to be relatively non-toxic to the manure-inhabiting predators it should not be routinely applied to the manure for fly control. Selective occasional applications to small areas ("spot treatment") having unusually high numbers of fly larvae would be satisfactory and have little or no effect on the overall predator population in a poultry house. Insecticide spraying of the manure in an entire house results in destruction of the predator population and only short-lived fly control (7-10 days) so that frequent repeated

applications become necessary (Axtell 1970b).

Selectivity can be partially achieved by the mode of insecticide use. Since flies rest on building surfaces and especially on the inside upper areas during the night, residual sprays on those resting surfaces can be used to kill adult flies with minimal risk to predators and parasites (Anderson and Poorbaugh 1964, Axtell 1970a). The little drift at the time of spraying will be generally insufficient to affect the manure-inhabiting predators. In the case of the pteromalid parasites there are no data, but it is likely that a part of the adult parasite population would be affected at the time of spraying, but that would be a temporary minor effect. The effects of the residual surface treatments on the parasites can only be determined after further behaviorial and susceptibility studies. There is presently no evidence that the residual treatments are reducing parasite populations. Presumably, since the parasites spend considerable time on the manure they largely avoid resting on the treated surfaces.

Another selective application approach is the use of toxicant-baits as adulticides. These are apparently not attractive to the parasites and are effective against the adult flies. The only possible risk to predators is from manure contamination resulting from indiscrimanant scattering of the bait rather than placing it in trays in areas of high fly activity.

The use of mist applications, from either portable equipment or permanently installed overhead equipment, for adult fly control should be of no risk to the predator population, but may be to the parasite population. Data are not available to judge the compatibility of frequent misting with the maintenance of high parasite populations.

AUGMENTATION OF PREDATOR AND PARASITE POPULATIONS

Naturally occurring populations of predators and parasites with proper manure management are often very high and the need for augmentation does not exist. In other situations, a sparse natural population may suggest the desirability of releasing additional predators and/or parasites, provided the manure management is satisfactory so that a suitable habitat exists for these biological control agents to operate effectively. Thus, the problem is: How do we determine whether or not to augment and with what species should we augment? We are far from having practical criteria for answering this. Indiscriminate releases of parasites and/or predators is often the practice now, but is unsatisfactory. An extensive review of augmentation with particular reference to flies has been published by Wiedhaas and Morgan (1976).

Factors to be evaluated before attempting augmentation are as follows. (1) Manure management.-Is the manure being maintained in as dry a condition as possible (as previously discussed) in order to maximize the populations of predators and parasites and minimize the suitability for fly breeding? (2) Existing predator and parasite populations.-What predators and parasites are present and how abundant are they? It may be pointless to augment with species that are already present and very abundant. (3) Fly populations.-Are the flies abundant enough to require further control attempts by augmentation? If the fly population is low, or there is liklihood of a decline due to seasonal or weather effects, then augmentation would be unnecessary. (4) Insecticide use.-Are insecticides being heavily used, including frequent manure treatment? If so, then the chances of successful augmentation are reduced. The insecticide use program may have to be modified, or greatly curtailed, prior to augmentation attempts.

To analyze these factors some practical measurements and observations are needed. Experienced personnel can observe the manure condition and recommend corrective action. The existing predator population can be determined by systematic examination of aliquots of manure spread onto a white surface or by extracting the fauna with Tulgren funnels. Many samples should be taken from different locations in a house. Counts of the macrochelid and uropodid mites and the Carcinops beetles are most important. Obviously some skill in recognizing those predators is required. The existing parasite population would be most effectively measured by exposing known-age (up to 24 house old) fly pupae by the pupal-bag technique as previously mentioned. This required some resources for fly rearing, however, and one must wait about 4 weeks for the parasites to emerge from the retreived pupae! The alternative of finding existing fly pupae and holding for parasite emergence could be used, but it is time-consuming and not very satisfactory. Measurement of the fly population can be deceptive unless a standarized method is used. The baited-jug method (Burg 1979, Rutz and Axtell 1979) is most practical and allows continuous sampling over several days between visits to a farm. The species of trapped flies can be readily determined. An alternative method is the "spot card" which is a white paper upon which resting flies leave defecation and regurgitation spots (Axtell 1970a). The numbers of spots indicate fly activity and this can be a continuous sample over several days. The disadvantages are that the species of flies cannot be determined and the positioning of the spot cards is critical. A third method, sticky fly ribbons, has been widely used and gives a sample of flies for which species can be determined but after 2 or 3 days the ribbon becomes ineffective due to being covered with flies and/or dust. The ribbons are inconvenient to handle and location is moderately critical. Determination of insecticide usage will depend

upon observations and the word of the poultry producer for records are seldom kept. The information on chemicals and dosage rates may be inaccurate.

With observations and data on the above factors at a particular poultry operation, it will be possible for an experienced person to make a subjective judgement as to whether or not augmentation is justified. Unfortunately, we lack sufficient data to quantitatively arrive at a conclusion. Eventually population modelling could provide the tools required. For the present, however, it is impossible to justify augmentation on the basis of quantitative data. Clearly, a subjective evaluation by experienced persons of the above factors is more justifiable than indiscriminate releases.

Not only is it difficult to justify augmentation in a given situation, it is further difficult to justify what species will be used. A great deal of attention has been give to using the parasite S. endius, but is it the "best" species and if so, what strain should be used? Clearly, it is risky to use strains shipped from elsewhere and which may not be adapted to the area of release, but this has been done. Should M. raptor be used and if so, what strain? It is a very abundant aggressive parasite and was a promising agent in our North Carolina experiments. Should predators such as M. muscaedomesticae and C. pumulio be released? They are apparently very effective suppressors of fly populations. What is the effect of long term colonization of parasites and predators on their effectiveness in the field? It's risky to rear parasites for many years and release them in the field (as has been done) without data to show that they are still effective.

Most important, one should avoid the implicit tendency to look for one parasite or one predator as the panacea for fly control. There has been a tendency to imply that a particular parasite species (S. endius, for example) can be the answer to fly control. This is unlikely to be the case. The use of multiple species of parasites and predators offers more promise for manipulation of the poultry manure ecosystem to minimize fly breeding.

SUMMARY

Predators and parasites associated with poultry manure are important biological control agents for the house fly and related filth flies. The development of maximum populations of these should be encouraged by proper cultural practices (manure management). A fly management program for poultry production must include an integration of biological, cultural and chemical (selective use of insecticides) methods. The practicality of this management approach has been demonstrated (Axtell 1970a, Legner and Dietrick 1974). Further, to be practical and acceptable to the poultry producers, the fly management program should

be a part of a Integrated Pest Management (IPM) program to include, in addition to flies, ectoparasites, rodents, diseases, and other pests of economic impact in these confined-animal production systems (Axtell 1981). Such a broad IPM program will allow skilled persons to conduct a pest monitoring program. This can include monitoring of biological control agents to facilitate rational decision-making about whether or not parasite or predator augmentation is justified. To more adequately use data from such a monitoring program, we need more data and efforts to develop reliable models of parasite, predator and fly populations (Ables, Shepard and Holman 1976, Wiedhaas et al. 1977, Wiedhaas and Morgan 1976). Potentially the use of parasite and/or predator releases to augment naturally occurring biological control of filth flies is promising as a part of an organized poultry IPM program, but considerable further research is essential.

REFERENCES

Ables, J. R., and Shepard, M.
1974. Responses and competition of the parasitoids Spalangia endius and Muscidifurax raptor (Hymenoptera: Pteromalidae) at different densities of fly pupae. Can. Entomol. 106: 825-830.

Ables, J. R.; Shepard, M.; and Holman, J. R.
1976. Development of the parasitoids Spalangia endius and Muscidifurax raptor in relation to constant and variable temperature: Simulation and validation. Environ. Entomol. 5: 329-332.

Anderson, J. R.
1965. A preliminary study of integrated fly control on northern California poultry ranches. Proc. California Mosquito Control Assoc. 33: 43-44.

Anderson, J. R., and Poorbaugh, J. H.
1964. Observations on the ethology and ecology of various Diptera associated with northern California poultry ranches. J. Med. Entomol. 1: 131-147.

Axtell, R. C.
1961. New records of North American Macrochelidae (Acarina: Mesostigmata) and their predation rates on the house fly. Ann. Entomol. Soc. Amer. 54: 748.
1963a. Acarina occurring in domestic animal manure. Ann. Entomol. Soc. Amer. 56: 628-633.
1963b. Effect of Macrochelidae (Acarina: Mesostigmata) on house fly production from dairy cattle manure. J. Econ. Entomol. 56: 317-321.
1964. Phoretic relationship of some common manure-inhabiting Macrochelidae (Acarina: Mesostigmata) to the house fly. Ann. Entomol. Soc. Amer. 57: 584-587.

Axtell, R. C.
1966. Comparative toxicities of insecticides to house fly larvae and Macrocheles muscaedomesticae, a mite predator of the house fly. J. Econ. Entomol. 59: 1128-1130.
1968. Integrated house fly control: Populations of fly larvae and predaceous mites, Macrocheles muscaedomesticae, in poultry manure after larvicide treatment. J. Econ. Entomol. 61: 245-249.
1969. Macrochelidae (Acarina: Mesostigmata) as biological control agents for synanthropic flies. Proc. Second International Congress of Acarology, 1967. Akademai Kiado, Budapest. p. 401-416.
1970a. Integrated fly control program for caged-poultry houses. J. Econ. Entomol. 63: 400-405.
1970b. Fly control in caged-poultry houses: Comparison of larviciding and integrated control programs. J. Econ. Entomol. 63: 1734-1737.
1981. Livestock integrated pest management (IPM): Principles and prospects. Proc. Symp. on Systems Approach to Animal Health and Production, Lexington, Kentucky.

Axtell, R. C., and Edwards. T. D.
1970. Hermetia illucens control in poultry manure by larviciding. J. Econ. Entomol. 63: 1786-1787.

Bouček, Z.
1963. A taxonomic study in Spalangia Latr. (Hymenoptera, Chalcidoidea). Acta Entomologica Musei Nat. Pragae 35: 429-511.

Burg, J. G.
1979. Some factors affecting baited-trap collections of house flies (Musca domestica) in caged-layer poultry houses. M.S. Thesis, N.C. State Univ., Raleigh, 60 p.

Coons, L. B., and Axtell, R. C.
1973. Sensory setae of the first tarsi and palps of the mite, Macrocheles muscaedomesticae. Ann. Entomol. Soc. Amer. 66: 539-544.

Farish, D. J., and Axtell, R. C.
1965. Sensory functions of the palps and first tarsi of Macrocheles muscaedomesticae (Acarina: Macrochelidae), a predator of the house fly. Ann. Entomol. Soc. Amer. 59: 165-170.
1971. Phoresy redefined and examined in Macrocheles muscaedomesticae (Acarina: Macrochelidae). Acarologia 13: 16-29.

Kogan, M., and Legner, E. F.
1970. A biosystematic revision of the genus Muscidifurax (Hymenoptera: Pteromalidae) with descriptions of four new species. Can. Entomol. 102: 1268-1290.

Legner, E. F.
1967. The status of Nasonia vitripennis as a natural parasite of the house fly, Musca domestica. Can. Entomol. 99: 308-309.
1971. Some effects of the ambient arthropod complex on the density and potential parasitization of muscoid Diptera in poultry wastes. J. Econ. Entomol. 64: 111-115.
1977. Temperature, humidity and depth of habitat influencing host destruction and fecundity of muscoid fly parasites. Entomophaga 22: 199-206.
Legner, E. F., and Dietrick, E. I.
1974. Effectiveness of supervised control practices in lowering population densities of synanthropic flies on poultry ranches. Entomophaga 19: 467-478.
Legner, E. F.; Moore, I.; and Olton, C. S.
1976. Tabular keys and biological notes to common parasitoids of synanthropic Diptera breeding in accumulated animal wastes. Entomol. News 87: 113-144.
Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.: Wiedhaas, D. E.; and Benton, A.
1975. Suppression of a field population of house flies with Spalangia endius. Science 198: 388-389.
O'Donnell, A. E., and Axtell, R. C.
1965. Predation by Fuscuropoda vegetans (Acarina: Uropodidae) on the house fly (Musca domestica). Ann. Entomol. Soc. Amer. 58: 403-404.
Peck, J. H.
1969. Arthropod predators of immature Diptera developing in poultry droppings in northern California. Part II. Laboratory studies on feeding behavior and predator potential of selected species. J. Med. Entomol. 6: 168-171.
Peck, J. R., and Anderson, J. R.
1969. Arthropod predators of immature Diptera developing in poultry droppings in northern California. Part I. Determination, seasonal abundance and natural cohabitation with prey. J. Med. Entomol. 6: 163-167.
Pfeiffer, D. G., and Axtell, R. C.
1980. Coleoptera of poultry manure in caged-layer houses in North Carolina. Environ. Entomol. 9: 21-28.
Rutz, D. A., and Axtell, R. C.
1978. Factors affecting production of the mosquito, Culex quinquefasciatus (= fatigans) from anaerobic animal waste lagoons. N.C. Agric. Expt. Sta. Tech. Bull. 256. 32 p.
Rutz, D. A., and Axtell, R. C.
1979. Sustained releases of Muscidifurax raptor (Hymenoptera: Pteromalidae) for house fly (Musca domestica) control in two types of caged-layer poultry houses. Environ. Entomol. 8: 1105-10.

Rutz, D. A., and Axtell, R. C.
1980a. House fly parasites (Hymenoptera: Pteromalidae) associated with poultry manure in North Carolina. Environ. Entomol. 9: 175-180.
1980b. Invasion and establishment of house fly, Musca domestica (Diptera: Muscidae) parasites (Hymenoptera: Pteromalidae) in new caged-layer poultry houses. J. Med. Entomol. 17: 151-155.
1981. House fly (Musca domestica) control in broiler-breeder poultry house by pupal parasites (Hymenoptera: Pteromalidae): Indigenous parasite species and releases of Muscidifurax raptor. Environ. Entomol.

Rutz, D. A.; Axtell, R. C.; and Edwards, T. D.
1980. Effects of organic pollution levels on aquatic insect abundance in field pilot-scale anaerobic animal waste lagoons. Mosquito News 40: 403-409.

Wiedhaas, D. E.; Haile, D. G.; Morgan, P. B.; and LaBrecque, G. C.
1977. A model to simulate control of house flies with a pupal parasite, Spalangia endius. Environ. Entomol. 6: 489-500.

Wiedhaas, D. E., and Morgan, P. B.
1976. Augmentation of natural enemies for control of insect pests of man and animals in the United States. Chpt. 14, p. 417-428. In: Ridgeway, R. L. and S. B. Vinson (eds.) Biological Control by Augmentation of Natural Enemies. Plenum Press, New York. 480 p.

Willis, R. C., and Axtell, R. C.
1968. Mite predators of the house fly: A comparison of Fuscuropoda vegetans and Macrocheles muscaedomesticae. J. Econ. Entomol. 61: 1669-1674.

Wise, G. U.
1970. Poecilochirus monospinosis new species. (Acarina: Parasitidae), a predator on the immature stages of the house fly. M.S. Thesis, N.C. State Univ., Raleigh, 56 p.

Wise, G. U., and Axtell, R. C.
1969. Predation by Poecilochirus sp. (Acarina: Parasitidae) on immature stages of the house fly, Musca domesticae. J. Elisha Mitchell Sci. Soc. 85: 122-123.

THE CONTRIBUTION OF *Spalangia endius* AND *Muscidifurax raptor* TO A STABLE FLY MANAGEMENT PROGRAM ON MACKINAC ISLAND, MICHIGAN: A QUESTION OF EFFORT

R. W. Merritt, E. F. Gersabeck and M. K. Kennedy

Department of Entomology, Michigan State University

INTRODUCTION

Mackinac Island is perhaps the oldest and most well known tourist attraction in the state of Michigan. It is located between Michigan's upper and lower peninsula in the Straits of Mackinac and encompasses 996 hectares with 13 kilometers of coastline.

The sole reliance on horse drawn carriages for transportation, automobiles are prohibited, is perhaps the most important feature of the island. Approximately 500-600 horses are needed to meet the transportation needs of over 750,000 tourists who visit the island each summer (Kennedy and Merritt 1980). However, the utilization of this type of low energy transportation is not without drawbacks. The most serious problem associated with the presence of large numbers of horses is the enormous accumulation of dung and subsequent increase in breeding sites for pestiferous flies. Despite efforts to dispose of both the horse manure and the prodigious amounts of garbage generated by the tourist trade, enough persists to allow the development of intolerable numbers of the house fly (*Musca domestica* L.) and the biting stable fly (*Stomxys calcitrans* [L.]).

Before 1945, fly control on Mackinac Island consisted largely of sanitation. In 1945, DDT was released by the U.S. Army for civilian use and was promptly tested on Mackinac Island for fly control. The results were very impressive; however, by 1949 the new "miracle drug" had lost its effectiveness. The island's fly population showed high levels of resistance to DDT. During the next 5 years, other chlorinated hydrocarbons (e.g. methoxychlor, chlordane, lindane, and dieldrin) were used in various combinations with the same results: the fly population quickly developed resistance to these chemicals

(Hoopingarner et al. 1966). This situation represented one of the earliest manifestations of the "pesticide treadmill" effect in the U.S.

Malathion was subsequently used until 1964 when the fly population again developed resistance (Hoopingarner and Krause 1968). Cygon (dimethoate) was used from 1964 until 1977 when resistance was suspected due to inadequate control (Kennedy and Merritt 1980). Resistance was verified in 1978 when Mackinac Island houseflies tested at the USDA Insects Affecting Man and Animals Laboratory in Gainesville, Florida were found to be 5 times more resistant to Cygon than a multiple resistant laboratory strain.

A second serious pest problem was detected on the island in the late 70's. An outbreak of the European fruit lecanium scale (Lecanium corni conplex) had seriously affected many of the shade and fruit trees located in or near the city and park (Kennedy 1977). Dieback of branches and a general decline in vigor was observed in trees infested with large numbers of scale. Our observations indicated that the dramatic increase in lecanium scale numbers on the island's trees was associated with weekly application of Cygon along city streets and horse trails for control of the filth-flies. This broad spectrum pesticide had eliminated the scale's natural enemies (parasites and predators) and allowed the pest to increase to damaging levels (Kennedy and Merritt 1980, Merritt et al. 1981).

Therefore, the island's previous fly control program caused three serious ecological problems: 1) increasing insecticide resistance in the target pest population, 2) a secondary pest outbreak, and 3) increasing human exposure to toxic chemicals. Clearly, alternatives to this type of unilateral control program were needed to avoid these adverse ecological consequences.

DEVELOPMENT OF AN INTEGRATED FLY MANAGEMENT PROGRAM

With the cooperation of the Mackinac Island State Park Commission and the Mackinac Island City Council, a pilot project was initiated in 1978. The major goal was to demonstrate that an integrated fly management program could be developed for Mackinac Island.

The program began with a survey of major fly breeding sites. Samples of manure, garbage and rotting vegetation from a variety of habitats were collected and examined for the presence of eggs, larvae, and pupae. Alsynite panels, which are highly attractive to adult stable flies (Williams 1973, Meifert et al. 1978), were covered with an adhesive substance and placed in areas where significant numbers of adults had been observed. Adult house fly populations were surveyed

using the grid method designed by Murvosh and Thaggard (1966) and supplemented by using sticky fly tapes.

Several situations were found to produce both stable and house flies on Mackinac Island. Horse manure mixed with hay and urine, especially when in contact with the ground, produced large numbers of flies in stables and corrals. Accumulated feed and moisture in cracks and crevices at the base of horse stalls also provided an excellent developmental medium. Manure wagons and boxes (areas of waste storage adjacent to stables) were not emptied on a regular basis, thus resulting in an optimum fly breeding medium. Several barns had seepage drains that allowed runoff from the stalls to drain into the yards. This resulted in small stagnant pools of waste materials that allowed larval breeding. In our survey, we found no parasitoids emerging from house or stable fly pupae on the island. It was likely that the past insecticide practice of spraying barns and stables with Cygon eliminated any parasitoids that might have been previously established.

Based on the results of the first year's pilot study, the following methods were recommended for the integrated management of pest flies on the island: 1) source reduction through sanitation, 2) composting manure to prevent larval and pupal development, 3) the placement of **Alsynite** panels coated with the insecticide permethrin around stable areas to increase adult mortality, 4) localized insecticide spraying at fly aggregation sites when ideal weather conditions favored the increased activity of adult flies, and 5) natural enemy releases.

NATURAL ENEMY RELEASES

In collaboration with USDA scientists in Gainesville, Florida, we decided to release parasitoids of the stable and house fly on the island. The two species selected Spalangia endius Walker and Muscidifurax raptor Girault and Saunders, are both pupal parasitoids of muscoid flies and have been shown to be effective in reducing numbers of house and stable flies in the field (Morgan et al. 1975, 1976; Weidhaas and Morgan 1977). S. endius was obtained through the USDA laboratory at Gainesville, Florida. Other pteromalids were obtained through commercial suppliers and subsequently determined to be M. raptor. In addition to periodic releases throughout the adult fly season, our major aim was to release parasitoids in the late fall and early spring to reduce the overwintering fly population.

Considerable effort was involved in the parasitoid release component of the fly management program. Table 1 shows a breakdown of the effort into various categories. Each category is accompanied by an estimate of the proportional amount of labor expended. Approximately one-half of the labor

was involved in determining fly developmental sites. We felt it was necessary to identify all major breeding areas within the flight range of the stable fly on the island. A map was divided into grids and representative sample sites from each section were examined for the presence of eggs, larvae or pupae. Once an estimate of fly production at each site was determined, a priority system for parasitoid releases was established based on fly pupal densities.

Many islanders associated hymenopteran parasitoids with hornets and yellow jackets. To overcome this "entomophobia," an educational program was initiated to increase the public's general knowledge of entomology before the parasitoid release was made. This effort, requiring approximately 25% of our time, took the form of weekly newspaper articles in addition to a radio and television program.

Table 1. Partitioning of effort in the parasitoid release program

Categories of Effort	% Effort
Determination of Sites	50
Education	25
Obtaining Access to Property	8
Training Personnel	8
Shipping, Handling, Distribution	2
Social and Political Interactions	2
Evaluation of Parasitoids	5
Rearing	?
Total	100
Proportion of Total Management Effort	30

Once the general public was exposed to information on parasitoid behavior and biology, access to property of individual cooperators for potential release sites had to be obtained (Table 1). Although many people claimed they understood the biology of the parasitoids, some thought the wasps would have to be controlled once the flies were gone while others were still concerned about being stung. In two instances, releases

were not made because of the cooperator's entomophobia of wasps.

Other social and political interactions involved maintaining a liason between the local City Council and State Park Commission to keep them informed on the parasitoid release program. Since these two groups supported and funded the program, they were concerned about their liability if the wasps started to sting tourists on the island. In addition, island administrators had to be convinced that biological control agents were not used in the same manner as conventional pesticides.

Although the shipping, handling and the distribution of parasitoids did not take a great deal of time (Table 1), it created some of our greatest problems. Shipping and receiving times had to coincide with the pest fly's biology to be effective. Also, because live material was being shipped, postal officials had to be alerted so that they would not inadvertently kill the insects through mishandling or storage. This was not a problem when parasitoids were shipped from the USDA laboratory in Gainesville, since containers were clearly marked that they contained live insects. However, in dealing with commercial suppliers, we encountered problems in guaranteed shipping dates, poor packaging and unmarked containers which made it impossible for postal authorities to alert us immediately after receiving a shipment.

Although commercial suppliers contracted with us to send S. endius, several shipments contained pure colonies of M. raptor. Also, suppliers claimed that 5-7 parasitoids could be expected to emerge from each fly pupa; however, for the above species, generally one and rarely two parasitoids have been reported to develop from a single pupa (Weidhaas and Morgan 1977, Weidhaas et al. 1977). As a consequence, shipments usually contained fewer numbers of parasitoids than specified in the original agreement.

The successful distribution of parasitoids in the field involved first placing wire mesh bags containing parasitized pupae in areas where they were not subject to human vandalism or animal curiosity. Evaluation of the introduced parasitoids required additional effort by trained technicians (Table 1). Pupal samples had to be collected from developmental sites, sorted, counted and held for parasitoid emergence in the laboratory. The information obtained on parasitization rates at different sites influenced the numbers of parasitoids released and the timing of subsequent releases.

CONCLUSIONS

The baseline data we collected in 1978 provided an index of adult stable fly densities during the fly season (Figure 1).

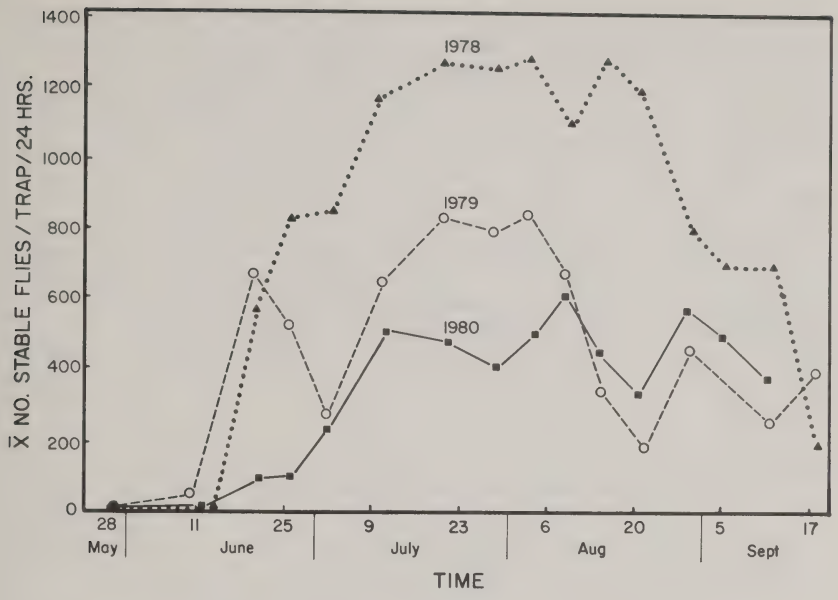


Figure 1. Changes in the mean number of S. calcitrans trapped on Mackinac Island during 1978-80. Each data point represents mean trap catch for 14 major problem areas.

In 1979, we estimated that source reduction through sanitation (without parasitoids) reduced the overall stable fly population by 35-37%. After the second year (1980), we achieved an additional 34-35% reduction in stable flies which we attributed to parasitoids as well as increased cooperation with sanitation efforts by island residents (Merritt et al. 1981) (Figure 1). House flies also declined 25-30% during the same time periods based on larval densities and cone trap counts. Sampling of muscoid pupae during the last year of the program revealed that 25-30% of those sampled were parasitized, whereas no parasitized pupae were found prior to the release program.

Our research indicated that pupal parasitoids of Diptera can produce significant mortality in isolated populations of pestiferous flies. However, the success of the parasitoid release required approximately 30% of our total management effort (Table 1). It is doubtful that the parasitoid release, in the absence of a total management program, would have produced equivalent results.

We felt that if parasitoids were to be a permanent and sucessful component of the integrated fly management program on the island, they would have to be reared locally. This

would have required a rearing facility, technical help and supplies for which the percent of effort could not be accurately determined (Table 1). However, a significant amount of effort was required, in addition to simply releasing parasitoids, to make them an integral component of the fly management program.

ACKNOWLEDGMENTS

We would like to thank Drs. R. S. Patterson and P. B. Morgan of the U.S.D.A. Insects Affecting Man and Animals Laboratory, Gainesville, Florida for supplying us with S. endius throughout the study. Research partially supported by USDA/SEA Competitive Animal Health Grant No. 59-2261-0-2-060-0.

REFERENCES

Hoopingarner, R. A.; Guyer, G. E.; and Krause, D. H.
1966. The Mackinac Island fly problem I. History of insecticide use and characterization of resistance. Mich. Agric. Expt. Sta. Quart. Bull. 48:559-64.

Hoopingarner, R. A., and Krause, D. H.
1968. The Mackinac Island fly problem II. Induced insecticide resistance. Mich. Agric. Expt. Sta. Quart. Bull. 50:281-4.

Kennedy, M. K.
1977. European fruit lecanium scale (Lecanium corni). Michigan State Univ. Coop. Ext. Insect, Disease and Nematode Alerts. No. 19:122.

Kennedy, M. K., and Merritt, R. W.
1980. Horse and buggy island. Nat. Hist. 89:34-41.

Meifert, D. W.; Patterson, R. S.; Whitfield, T.; LaBrecque, G. C.; and Weidhaas, D. E.
1978. Unique attractant-toxicant system to control stable fly populations. J. Econ. Ent. 71:290-2.

Merritt, R. W.; Kennedy, M. K.; and Gersabeck, E. F.
1981. Integrated pest management of nuisance and biting flies in a Michigan resort: An answer to secondary pest outbreaks. In: (G. W. Frankie and C. S. Koehler, eds.), Urban Entomology. Praeger Scientific, New York (In press).

Morgan, P. B.; Patterson, R. S.; and LaBrecque, G. C.
1976. Controlling house flies at a dairy installation by releasing a protelean parasitoid Spalangia endius Walker. J. Georgia Entomol. Soc. 11:39-43.

Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Weidhaas, D. E.; and Benton, A.
1975. Suppression of a field population of houseflies with Spalangia endius. Science 189:388-9.

Murvosh, C. M., and Thaggard, C. W.
1966. Ecological studies of the house fly. Ann. Entomol. Soc. Amer. 59:533-47.

Weidhaas, D. E.; Haile, D. G.; Morgan, P. B.; and LaBrecque, G.C.
1977. A model to simulate control of house flies with a pupal parasite, Spalangia endius. Environ. Entomol. 6:489-500.

Weidhaas, D. E., and Morgan, P. B.
1977. Augumentation of natural enemies for control of insect pests of man and animals in the United States, pp. 417-428. In: Ridgway, R. L. and S. B. Vinson, eds.). Biological control by augmentation of natural enemies. Plenum Press, New York.

Williams, D. F.
1973. Sticky traps for sampling populations of Stomoxys calcitrans. J. Econ. Ent. 66:1279-80.

MASS RELEASE OF PUPAL PARASITES FOR CONTROL OF STABLE FLIES AND HOUSE FLIES IN CONFINED FEEDLOTS IN NEBRASKA

D. A. Stage and J. J. Petersen

University of Nebraska and U.S. Department of Agriculture

The sale of hymenopterous parasites for the control of insect pests has been underway for many years. However, recently the numbers of commercial distributors promoting pteromalid pupal parasites for house and stable fly control have increased noticeably. The management of a number of dairies and confined livestock operations throughout the midwest have been promised effective fly control either through simple application of these wasps or through a combination of cultural control (sanitation), limited pesticide usage and parasite releases. The effectiveness of these parasites is being promoted without evidence that a given parasite species or application procedure is effective under confined livestock conditions. The only 'proof' of effectiveness commercial parasite producers can draw upon is testimonials of some feedlot operators. These favorable testimonials may be the result of wishful thinking, improper observation, better sanitation or favorable environmental conditions occurring concurrently with the scheduled release programs.

Though documented research is not available on the effectiveness of pteromalid wasps for the control of filth breeding flies on the larger confined livestock feedlots (in excess of 100 animals), a number of studies have been made in confined and semiconfined poultry houses (Legner and Brydon 1966, Legner and Dietrick 1974, Olton and Legner 1975, Rutz and Axtell 1979). A review of the literature reveals that at least seven species (4 genera) of the parasites have been employed in controlled release programs. However, only two, Spalangia endius Walker and Muscidifurax raptor Girault and Sanders, have been intensively tested. Morgan et al. (1975a,b) reported complete eradication of house fly populations in poultry houses in 30-35 days in two separate trials using S. endius. Also, they reported 100% parasitism on two small dairies with S. endius (Morgan et al. 1976, Morgan and Patterson 1977). Significant but less dramatic levels of parasitism were achieved in poultry

houses in California using a combination of 4 pteromalid species (Legner and Dietrick 1972, 1974) and North Carolina using M. raptor (Rutz and Axtell 1979).

This study was conducted to evaluate the effectiveness of commercially reared parasites when released as recommended by a distributor who rears and sells these parasites for the control of filth breeding flies on dairies and livestock feedlots. Aims of the study were to measure the effect the parasites had on pupae of house and stable flies on two confined feedlots; and to evaluate the numbers, species, and quality control of the parasitic wasps supplied by the vendor.

MATERIALS AND METHODS

A representative of a company, commissioned to supply the parasites for the study, scouted six feedlots in Cuming County, Nebraska in April, 1980 and made recommendations for a parasite release program on each lot. Four of the lots were selected for the study becasue of similarities in size, number of cattle and potential fly breeding areas. Then two of these lots were randomly selected for parasite releases and two to serve as controls.

One of three species of parasites [Nasonia vitripennis (Walk.), S. endius or M. raptor] was provided for release each week from May 13 to October 11, 1980. The species and numbers of parasites scheduled for a given release were determined by the distributor. The wasps were shipped by U.P.S. as parasitized house fly pupae in wood shavings. Immediately on arrival, the pupae were counted and checked for early parasite emergence. Then random samples of 100 pupae were separated and held to determine the extent of parasite emergence and species identification. Parasites were released directly into known fly breeding areas on the two release lots within 24 hours after the first emergence of parasites. Care was taken in the release of the parasitized pupae to provide protection from direct sunlight and desiccation.

The incidence of pupal parasitism was measured by two methods. First, from May to August ten aluminum pans containing moist vermiculite and 20-40 recently pupated stable flies from a laboratory colony were buried 2-6 cm in fly breeding areas on each of the four feedlots. After five days of exposure, the pupae were returned to the laboratory, floated from the debris, and placed individually into gelatin capsules and held at 27 $\pm$ 2°C and ca. 70% RH for emergence of flies and parasites. Second, random populations of 100-300 wild house fly and stable fly pupae were collected on a weekly basis from May-October from all four feedlots. Care was taken to avoid making excessive collections from sites of dense pupal concentrations to insure an adequate area-wide sample. The pupae were returned to the laboratory, separated out, identified to species and

handled as previously described. The percentage parasitism was calculated by dividing the number of pupae which yielded parasites by the total number of pupae yielding flies or parasites and multiplying by 100.

Adult house and stable fly populations were monitored weekly from May through November by using traps made of Alsynite panels supported one meter above the ground with a surface area of 3300 cm^2. Six traps were placed around the perimeter of each feedlot. Flies were collected weekly by covering the panels of each trap with a plastic sleeve and then applying "Tack Trap." After 24 hours, the numbers of trapped house and stable flies were counted and recorded. Stable fly populations were also monitored by making standard weekly leg counts on 10 randomly selected calves per lot.

RESULTS AND DISCUSSION

The frequency of shipment of a given parasite species indicated that the distributor supplied species based on their availability rather than their potential for controlling fly populations (Table 1). Also, quality control checks showed shipments of M. raptor and S. endius were well below those promised by the supplier. Shipments of M. raptor and S. endius produced only 48 (40-55)% and 31 (16-48)% of the promised yield of parasites, respectively. However, the numbers of N. vitripennis usually exceeded the numbers promised by 3-4 times and as much as seven times. This oversupply of N. vitripennis undoubtedly resulted when procedures used for estimating numbers of M. raptor and S. endius were used for N. vitripennis, a parasite that typically produces multiple parasitisms of its host.

The level of parasitism as determined by laboratory reared stable fly pupae averaged 2.1 percent for the two control lots and 12.7 percent for the two release lots (Table 2). Levels of parasitism were similar but higher in wild pupae averaging 9.8 percent for the control lots and 19.8 percent for the release lots. Parasitism on all four lots showed a marked increase in September in both laboratory and wild pupae. Again, increases were greater on the release lots than on the control lots, with parasitism of wild pupae on control and release lots reaching 43.3 and 73.4 percent, respectively.

In all cases wild pupae produced higher levels of parasitism than laboratory pupae. The lower levels of parasitism may have resulted from laboratory pupae being less attractive to the parasites for a number of unknown reasons. However, since the parasites require 3-4 weeks to complete development, parasitized hosts tend to accumulate in the environment giving the appearance of higher levels of parasitism in wild pupae.

Table 1.--Numbers and species of parasitic wasps supplied by a commercial distributor for fly control on two confined livestock feedlots in Nebraska

Week	Number of parasites per lot: Shipped number	Number of parasites per lot: Received and released	Species
1	80,000	44,200	M. raptor
2	80,000	31,700	M. raptor
3	80,000	5,500	N. vitripennis
4	80,000	165,100	N. vitripennis
5	60,000	160,500	N. vitripennis
6	60,000	188,000	N. vitripennis
7	60,000	215,600	N. vitripennis
8	60,000	30,200	M. raptor
9	50,000	20,400	S. endius
10	50,000	9,100	S. endius
11	50,000	24,000	S. endius
12	50,000	14,900	S. endius
13	40,000	12,000	S. endius
14	40,000	279,000	N. vitripennis
15	40,000	6,600	S. endius
16	40,000	156,000	N. vitripennis
17	40,000	198,700	N. vitripennis
18	40,000	162,000	N. vitripennis
19	40,000	131,500	N. vitripennis
20	40,000	128,400	N. vitripennis
21	60,000	165,100	N. vitripennis
22	60,000	288,600	N. vitripennis
Totals	1,200,000	2,437,100	

The parasites collected from the pupal samples are now in the process of being identified. Preliminary data indicate that a native species, Spalangia nigroaenea Curtis, was the predominant species throughout the season on all four feedlots and was mainly responsible for the significant increases in levels of parasitism observed in late summer and early fall. Although N. vitripennis was released in large numbers (2,200,000 per lot) over the 22 week period of the study, only one specimen of this species was recovered from ca. 12,000 pupae sampled from the release lots.

Adult stable fly population trends were generally the same on all four lots; they first appeared the last week in May with populations reaching a peak the first week in July. These populations declined through the remainder of July and early August, and built up to a second but lower peak early in

Table 2.--Parasitism of house fly and stable fly pupae by pteromalid wasps on four (two release and two control) confined livestock feedlots

Pupae	Percentage parasitism			
	Release lots		Control lots	
	1	2	1	2
Laboratory stable flies	4.7	20.6	1.5	2.7
Wild stable flies	14.8	24.3	5.0	12.6
Wild house flies	13.8	27.3	6.6	18.2

September followed by steady decline throughout the remainder of the season. The peak populations of stable flies based on trap counts were lower on the release lots than on the control lots. Leg counts for stable flies proved to be somewhat erratic due to the weather conditions and inherent inconsistencies in the counting method and proved inconclusive.

The same general trends occurred for house fly populations on all four lots; populations increased throughout the summer and peaked during the third week in September. However, unlike the stable flies, house fly populations were higher on the release lots than on the control lots.

Although adult stable fly populations were lower on the release lots, this reduction could not be attributed to the released parasites. The periods of reduced fly populations did not always correspond with observed increases in the levels of parasitism. Furthermore, since S. nigroaenea was the predominant parasite species, much of the parasitism observed on the release lots could not be attributed to the releases. Nasonia vitripennis has been shown to be ineffective for the control of flies under natural conditions. This work strongly supports the warnings by Legner (1967) and Wylie (1958) against using N. vitripennis as a biological control agent for filth breeding flies. Of the three release species, M. raptor was the most frequently recovered. However, this species was also collected in substantial numbers on the control lots making it difficult to assess the effect of releases of fly populations especially in light of the low numbers that were released (106,000 per lot).

The practical use of pupal parasites for the control of filth breeding flies in the midwest remains to be demonstrated. Much needed background information on the impact of natural parasite populations of flies on confined feedlots must be obtained to properly evaluate parasite release programs. Also, research is needed to determine the most effective species of

parasite, the optimal times for releases, and the optimum numbers of parasites to release for feedlots in this area.

Failure of this release program does not necessarily demonstrate the ineffectiveness of pupal parasites for fly control. It does suggest that parasite distributors may be uninformed of the biology of the parasites they are selling or have ignored biological information when making recommendations to owners. It is evident that much remains to be learned before parasitic wasps can be recommended as an effective control for filth breeding flies on confined feedlots in the midwest.

REFERENCES

Legner, E. F.
1967. The status of Nasonia vitripennis as a natural parasite of the house fly, Musca domestica. Can. Entomol. 99: 308-309.

Legner, E. F., and Brydon, H. W.
1966. Suppression of dung--inhibiting fly populations by pupal parasites. Ann. Entomol. Soc. Am. 59(4): 638-651.

Legner, E. F., and Dietrick, E. J.
1972. Inundation with parasitic insects to control filth breeding flies in California. Proc. Calif. Mosq. Control Assoc. 40: 129-130.
1974. Effectiveness of supervised control practices in lowering population densities of synanthropic flies on poultry ranches. Entomophaga 19(4): 467-468.

Morgan, P. B., and Patterson, R. S.
1977. Sustained releases of Spalangia endius to parasitize field populations of three species of filth breeding flies. J. Econ. Entomol. 70: 450-452.

Morgan, P. B.; Patterson, R. S.; and LaBrecque, G. C.
1976. Controlling house flies at a dairy installation by releasing a protelean parasitoid Spalangia endius (Hymenoptera: Pteromalidae). J. Ga. Entomol. Soc. 11: 39-43.

Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Benton, A.; and Whitfield, T.
1975a. Rearing and release of the house fly pupal parasite Spalangia endius Walker. Environ. Entomol. 4: 609-611.

Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Weidhass, D. E.; and Benton, A.
1975b. Suppression of a field population of house flies with Spalangia endius. Science 189: 388-389.

Olton, G. S., and Legner, E. F.
1975. Winter inoculative releases of parasitoids to reduce house flies in poultry manure. J. Econ. Entomol. 68(1): 35-38.

Rutz, D. A., and Axtel, R. C.
1979. Sustained releases of Muscidifurax raptor (Hymenoptera: Pteromalidae) for house fly (Musca domestica) control in two types of caged--layer poultry houses. Environ. Entomol. 8: 1105-1110.

Wylie, H. G.
1958. Factors that affect host finding by Nasonia vitripennis (Walk.) (Hymenoptera: Pteromalidae). Can. Entomol. 90: 597-608.

USE OF PACHYCREPOIDEUS VINDEMIAE TO CONTROL FLY BREEDING IN POULTRY COOPS AND CATTLE STALLS

Lawrence G. Pickens

U.S. Department of Agriculture

Although Pachycrepoideus vindemiae (Rondani) is not presently being used to any large extent as a biocontrol agent, our experiments with it may be of interest to anyone who is working with other pteromalids.

We reared and released ca. 5700 wasps each week in small poultry coops and in a screened-in calf box stall in 1975 and measured the percentage of the collected house fly and Fannia spp pupae which were parasitized and the percentage reduction in the number of adult house flies, stable flies and Fannia spp in the wasp-release areas compared with an untreated "check" area (Pickens et al. 1975).

Although each female wasp killed an average of 4 house pupae per day for 15 days (a total of 60 pupae/♀) in the laboratory, they only averaged 0.4-1.3 pupae/♀ per day in the field test (a total of ca. 12 pupae per♀). Thus, an average of 5700 wasps per week (633 wasps/m^2 of bedding) only reduced the house fly population in a box stall by ca. 40%. However, a release of ca. 4800 wasps (1600/m^2 of feces) per week in small (3 x 3 x 3m) poultry coops reduced the adult populations of house flies by 82-90%, of stable flies by 90%, and of Ophera leucostoma (black garbage fly) by 84% after 70 days. Since O. leucostoma is a predator on house fly larvae, this could be detrimental.

Probably the wasps were less effective in the box stall than in the poultry coop because of the greater area in which host pupae were dispersed.

In separate tests of hunting efficiency, we found that the percentage of host pupae killed decreased by 10% for every 3 square meters of area to be searched.

The effectiveness of the wasp as a control agent was also affected by temperature, with the length of the immature period varying from 14 to 28 days and with up to 70% of the newly-released wasps leaving the coop at high temperatures (>29°C).

As a result, it would require an estimated daily population of 2800 wasps per square meter of feces to achieve complete control of house flies breeding in poultry feces or 700 wasps/day/m^2 of bedding in a box stall in one fly generation.

Since the adult wasps usually died in 7 days if no host pupae were available, mass-releases of the wasps during periods of low fly numbers may not be worthwhile unless dead fly pupae are placed at the release sites to serve as surrogate hosts.

Large numbers of female wasps per unit area reduced the wasp female to male progeny ratio from 3.5:1 to 1.1:1.

House fly pupae which were frozen when they were 2 days old produced 20% more wasps than did living pupae. The adult wasps were attracted to living or dead house fly pupae and to glass plates which were sprayed with a petroleum ether extract of house fly pupal cases.

When dead house fly pupae were placed in poultry coops 3 times per week (1500 pupae/m^2 of feces), released wasp populations were able to maintain themselves as well as did wasp populations which were augmented by 1000 wasps/m^2 of feces 3 times a week (Pickens and Miller 1978).

Wasps which were released for 1 month only and which were not supplied with surrogate hosts failed to maintain themselves for more than 8 weeks.

Finally, although many of the wasps dispersed to a poultry coop which was 100 m from the coop where they were released, very few of them were found in a coop 1 km away from the release site, and very few pupae were parasitized when they were in dark areas of buildings which had windows.

REFERENCES

Pickens, L. G.; and Miller, R. W.
1978. Using frozen host pupae to increase the efficiency of a parasite-release program. The Florida Entomol. 61: 153-158.

Pickens, L. G.; Miller, R. W; and Centala, M. M.
1975. Biology, population dynamics, and hose finding efficiency of Pachycrepoideus vindemiae in a box stall and a poultry house. Environ. Entomol. 4: 975-979.

NATURAL PARASITE LEVELS IN HOUSE FLIES, STABLE FLIES, AND HORN FLIES IN FLORIDA

J. F. Butler and R. Escher, University of Florida, and
J. A. Hogsette, U.S. Department of Agriculture

INTRODUCTION

Numerous ecological studies made over the past five years on filth breeding flies have allowed us to accumulate data on parasitism of horn flies, house flies, and stable flies. These studies include works by Greer (1974), Wilkerson (1974), Escher (1977), Kramer (1975), and Hogsette (1980). Identification of pupal parasites was verified by Dr. E. Grizelle, U. S. National Museum of Natural History, Washington, D.C.

Data were collected on species of parasites present, percent parasitism, the sex ratios of adult parasites, and when possible, the seasonal occurrence for the parasites under study.

These studies required the development of techniques for the recovery of pupal parasites of known age from larval media, as well as standardization of sampling techniques, so that field surveys of parasitized pupae could be made. In most cases an estimate of the time of development to pupal eclosion had to be made, so that field exposure would allow time for normal parasitism to occur. In this way samples could be taken before adult flies emerged but after pupal exposure to parasitism. The time required for horn fly [*Haematobia irritans* (L.)] pupal exposure was based on a developmental rate formula established by Wilkerson (1974). This formula allowed calculation of developmental time based on the mean daily temperature and allowed for parasitized pupae to be collected throughout the year.

HORN FLY PARASITISM

Whole cow pats or artificial cow pats which had been seeded with horn fly eggs or first instar larvae were placed near a pasture with cattle. Artificial pats were made from fresh cow

manure which had been frozen for 48 hours to kill extraneous organisms. Natural pats were either seeded with eggs or larvae or exposed to natural horn fly oviposition. The pats were marked and recovered for extraction of pupae at a time calculated to precede eclosion of adult horn flies (Escher 1977).

Pupae were recovered from pats by a flotation technique (Greer 1974). Recovered pupae were then held at a constant temperature (26.6 ± 1°C) until emergence of the parasite occurred. Those pupae failing to hatch were dissected to determine if parasitism had occurred.

Adult fly populations on the pastured animals in the experimental area were counted as the number of flies per one-half animal; ten animals were selected at random and counted for each count date. Temperature and rainfall data were supplied by a nearby climatological station of the University of Florida Agronomy Department.

Horn fly populations in the test area varied between the different pastures used in the study, but had similar bimodal

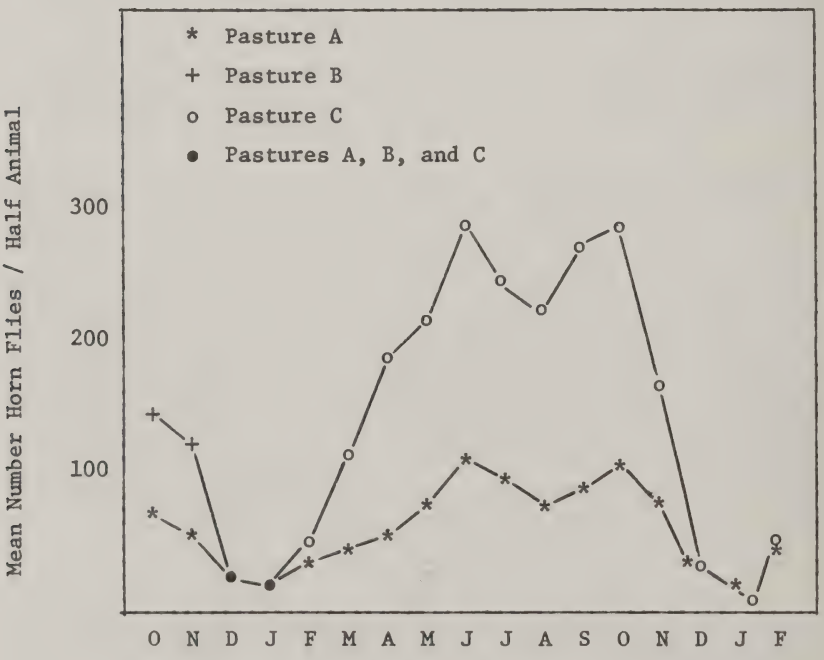


Figure 1.--Horn fly levels for animals on Pastures A, B, and C from 1975 to 1976

peaks of abundance in May through June and September through October (figure 1). Pasture C was more typical of normal fly populations observed on beef cattle in Central Florida with peak numbers reaching 300/half animal.

The percent parasitism of pupae collected from sites near these pastures is presented in figure 2. Both percent parasitism (figure 2) and fly population (figure 1) declined as rainfall increased and temperature decreased in October and November. Percent parasitism peaked in June at over 20% of the fly pupae.

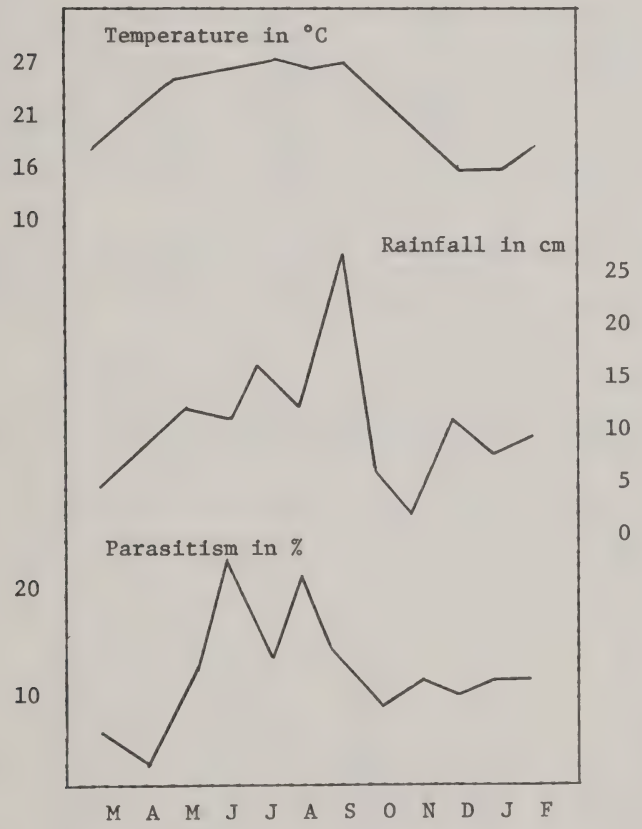


Figure 2.--Comparison of total parasitism rate by hymenopterous parasites on horn fly pupae with the average monthly rainfall and temperatures recorded between 1975 and 1976.

Horn fly pupation occurred both within the aged manure pat and in the sand under the manure pat. Parasitism of horn fly pupae recovered from the manure pat is presented in table 1, while those recovered from the sand under the pat is presented in table 2. Parasitism in both sites was highest in May, June, and August. Both pupae and parasites were recovered from the sand under the pat (table 2), showing that horn flies pupate in the sand under the pat and parasites can find them, but at a reduced rate. It is evident that although both parasitized and unparasitized pupae are found in both sites, they are about four times as abundant within the pat, but much less likely to be parasitized beneath the pat.

When seeded pats were evaluated for survival of horn flies, the bimodal peak was again seen. The percent pupal survival from larvae (figure 3) seldom exceeded 40%, with the exception of May through June, September, and November. This also corresponds to the adult bimodal peak (figure 1). Further work is necessary to correlate pupal survival and percent parasitism to environmental conditions, although in this study rainfall and temperature appear important.

Identification of parasites recovered from horn fly pupae is given in table 3. Ten different species were recovered with *Spalangia* being the most common genus.

Table 1.--Percent parasitism of horn fly pupae by hymenopterous parasites collected from pats in Pastures A, B, and C between 1975 and 1976.

Collection Date	Pats	Puparia	Parasites	% Parasitism
March	14	652	40	6.1
April	38	885	17	1.9
May	32	1536	159	10.4
June	24	1988	389	19.6
July	32	2314	228	9.9
August	40	1382	224	17.7
September	24	1486	127	8.6
October	28	1368	90	6.6
November	42	2769	258	9.3
December	6	276	21	7.6
January	25	446	41	9.2
February	16	1830	162	8.8
TOTALS	321	16932	1776	10.5

Table 2.--Percent parasitism of horn fly pupae recovered from the sand beneath the artificial pats located in Pastures A and C between 1975 and 1976.

Collection Date	Pats	Puparia	Parasites	% Parasitism
March	14	153	0	0.0
April	38	219	0	0.0
May	32	466	12	2.6
June	24	561	27	4.8
July	32	974	5	0.5
August	40	535	11	2.1
September	24	747	7	0.9
October	12	221	3	1.4
November	16	310	4	1.3
December	4	9	0	0.0
January	8	18	0	0.0
February	16	454	14	3.1
TOTALS	260	4666	83	1.8

Table 3.--Species of horn fly pupal parasites collected from natural and artificial cow pats in North Central Florida. Identifications were verified by Dr. Eric Grizelle, U. S. National Museum of Natural History, Washington, D.C.

1. *Spalangia haematobiae* Ashmead *
2. *S. cameroni* Perkins *
3. *S. nigra* Latreille
4. *S. nigroaenea* Curtis
5. *S. endius* Walker
6. *Muscidifurax raptor* Girault and Saunders
7. *Pachycrepoides vindemmiae* (Rondani)
8. *Aphaereta pallipes* (Say) Braconidae
9. *Pseudeucoilia* sp.
10. *Trichopria* sp. (Diapriidae)

* Most important species

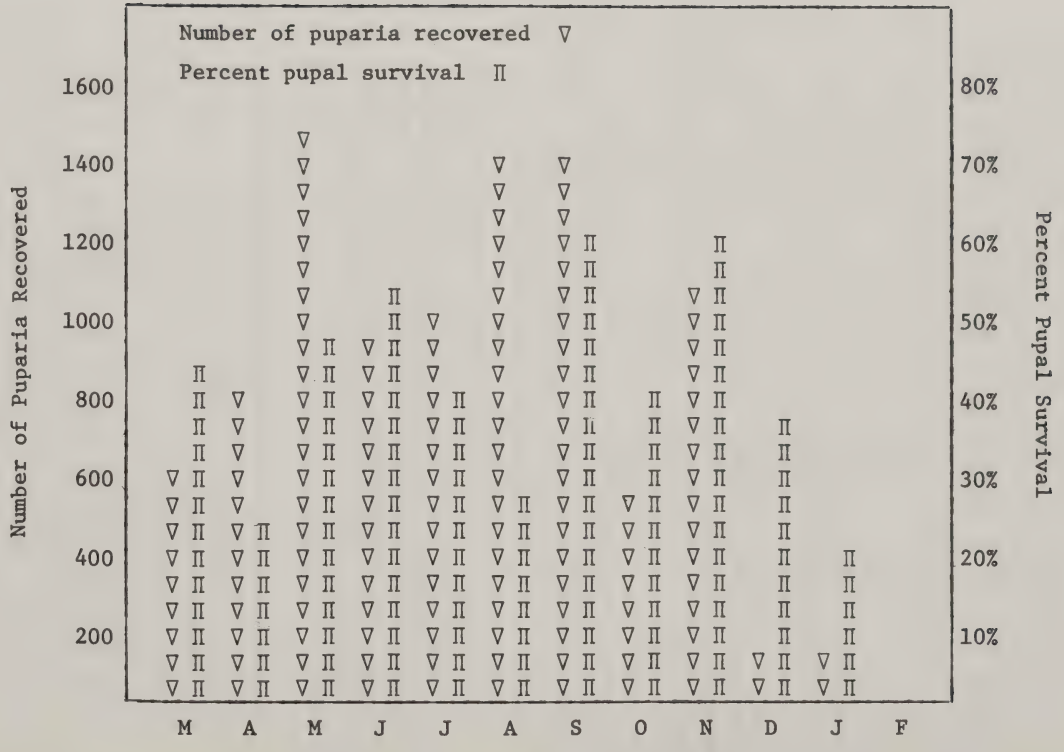


Figure 3.--Survival of horn fly larvae to pupation placed in Pastures A and C between 1974 and 1976 at the rate of 100 larvae per artificial pat.

The percent parasitism by species of pupal parasite is given in table 4. Seasonally, *Spalangia cameroni* was found at the highest rate during the warmest months of May through September. During the cooler months, *S. haematobiae* was seen as an important species. *S. nigroaenea* was the third most important.

Spalangia cameroni Perkins accounted for 44.6% of the total number of parasitized pupae collected. *S. cameroni* was also found to be the most common parasite of horn fly pupae in Mississippi by Combs and Hoelscher (1969). Eighty percent of all pteromalids, which represented 94% of the parasites collected by them, were identified as *S. cameroni*. This species was not reported as a parasite of horn flies by Lindquist (1936) in Texas, Depner (1968) in Alberta, or by Thomas and Morgan (1972) in Missouri.

Spalangia haematobiae Ashmead, the second most common parasite collected in this investigation, accounted for 23.6% of all parasitized pupae collected. *Spalangia haematobiae* was collected by Thomas and Morgan (1972) from horn fly puparia in all three years of their investigation of horn fly pupal parasites in Missouri. It was found to be a regular parasite of horn fly pupae (0.23%), but was much less common than *S. nigra* (32%) and *S. nigroaenea* (27%). *S. haematobiae* was reported by Depner (1968), but this determination was changed to *Spalangia subpunctata* Forester by Peck (1974). *S. haematobiae* was not reported from Texas by Lindquist (1936), but later examination of some of this material by Boucek (1963) revealed that some of the specimens were *S. haematobiae*. *S. haematobiae* was not reported from Mississippi by Combs and Hoelscher (1969). According to Boucek (1963), *Spalangia haematobiae* has only been recovered from horn fly puparia in the United States.

Spalangia nigra Latreille accounted for 9.1% of the parasites identified in this investigation. Thomas and Morgan (1972) found *S. nigra* to be the most common pupal parasite of the horn fly in Missouri. *S. nigra* was not recovered by Combs and Hoelscher (1969) in Mississippi, by Depner (1968) in Alberta, or by Lindquist (1936) in Texas.

Spalangia nigroaenea Curtis accounted for 7.5% of the parasites identified in this investigation. *S. nigroaenea* was the second most common parasite of horn fly pupae collected by Thomas and Morgan (1972) in Missouri and Combs and Hoelscher (1969) in Mississippi. There it accounted for 14% of the 94% of the parasites collected and identified as pteromalids. *S. nigroaenea* was not reported from Alberta by Depner (1968) or by Lindquist (1936) in Texas, although it was reported from Texas as a parasite by Bishopp (1913).

Spalangia endius Walker appears to be a very rare pupal parasite of the horn fly in this area. It accounted for only 3.5%

Table 4.--Percent parasitism of horn fly pupae due to each species of hymenopterous parasite per month. Pupae were collected from Pastures A and C between 1975 and 1976.

Collection Date	*S.c.*	*S.h.*	*S.n.*	*S.nig.*	*S.e.*	*M.r.*	*P.v.*	*A.p.*	*T*	*P*	U.k.
March	1.54	0.24	0.24	0.18	0.06						
April	0.41	0.06	0.12			0.24	0.12			0.06	
May	3.84	0.18	0.24	1.00	0.30	0.24	0.30				3.31
June	12.46	0.71	0.71	0.71	4.72			0.06	0.18		0.83
July	7.68	2.83	0.89	0.95	0.06					0.06	0.59
August	7.32	3.72	2.60	0.12						0.06	0.59
September	3.60	2.24	0.65	0.18							0.53
October	0.41	0.71									0.12
November	1.77	2.24	1.53		0.06				0.06		0.24
December	0.59	0.47		0.18							
January				0.24							
February	0.83	5.43	0.77	0.53					0.06		1.90

S.c. *Spalangia cameroni*
S.h. *Spalangia haematobiae*
S.n. *Spalangia nigroaenea*
S.nig. *Spalangia nigra*
S.e. *Spalangia endius*
M.r. *Muscidifurax raptor*
P.v. *Pachycrepoideus vindemminae*
A.p. *Aphaereta pallipes*
T *Trichopria* sp.
P *Pseudeucoila* sp.
U.k. Unidentifiable immatures

of the parasites identified in this survey. *S. endius* has been reported as a horn fly pupal parasite by Lindquist (1936) in Texas. In that survey *S. endius* accounted for more than 95% of all horn fly pupal parasites collected. *S. endius* was not reported as a horn fly pupal parasite in Missouri (Thomas and Morgan 1972), Mississippi (Combs and Hoelscher 1969), or in Alberta (Depner 1968).

A *Trichopria* sp. accounted for 0.68% of the pupae parasitized in this investigation. A *Trichopria* sp. was reported as a horn fly pupal parasite by Thomas and Morgan (1972) in Missouri, where it accounted for 0.02% of the parasites collected. Combs and Hoelscher (1969) also recovered a *Trichopria* sp. from 2% of the horn fly puparia which they collected in Mississippi. Depner (1968) in Alberta and Lindquist (1936) in Texas did not report *Trichopria* sp. as a pupal parasite of the horn fly.

Muscidifurax raptor Girault and Saunders was rarely collected in this investigation and accounted for only 0.45% of the parasites collected. Depner (1968) reared *M. raptor* from horn fly puparia in Alberta, where it was the second most common of nine genera of horn fly pupal parasites collected. *M. raptor* was also reared from horn fly puparia collected by Thomas and Morgan (1972) in Missouri, where it accounted for 4% of all parasites collected. *M. raptor* was not recovered in Mississippi by Combs and Hoelscher (1969) or in Texas by Lindquist (1936).

Pachycrepoideus vindemmiae (Rondani) which represented 0.39% of the parasites collected in this survey has never before been reported as a horn fly pupal parasite.

A *Pseudeucoilia* sp. represented 2% of the parasites collected by Thomas and Morgan (1972) in Missouri and 1% of those reared by Combs and Hoelscher (1969) in Mississippi. A *Pseudeucoilia* sp. was a very rare pupal parasite in the present investigation and accounted for only 0.11% of the parasites collected. Depner (1968) in Alberta and Lindquist (1936) did not report a *Pseudeucoilia* sp. in their investigations.

Aphaereta pallipes (Say) accounted for 1% of all parasites reared from horn fly puparia collected by Thomas and Morgan (1972) in Missouri and also by Combs and Hoelscher (1969) in Mississippi. In the present investigation *A. pallipes* represented only 0.06% of the 1776 pupal parasites collected. *A. pallipes* was not reported by Depner (1968) from Alberta or by Lindquist (1936) from Texas.

From these data it appears that horn fly parasitism is predominantly by *S. cameroni*, *S. haematobiae*, and *S. nigra*. All other species were found less frequently. *S. cameroni* was found at high rates early in the season (May through September), while *S. haematobiae* had lower parasite rates but were found later in the season (July through September, November, and February).

STABLE FLY PARASITISM

A single trial was conducted which involved natural parasitism of stable fly pupae by pupal parasites. In this trial, artificial containers with wood chip/paper mill extract were seeded with first instar stable fly larvae (*Stomoxys calcitrans*). These containers were exposed to field populations of parasites on the campus of the University of Florida. Upon emergence of parasites only one species, *Spalangia nigra* Latreille, was recovered with about 1% of the pupae parasitized.

PUPAL PARASITISM RATES AS AFFECTED BY METHOPRENE

Trials were conducted on stable flies and horn flies to determine the effect on natural parasitism of media treated with methoprene (ZR515 or Altosid) (Butler unpublished research report 1973).

Wood chip/paper mill extract media, which was producing large numbers of stable flies under natural conditions, was brought to the laboratory, artificially infested with *Stomoxys calcitrans*, and treated with methoprene at 10 ppm (table 5).

This stable fly medium produced a parasitism rate of 14-18%, as compared to 1% for untreated media. This JH compound is characterized by interruption of pupal eclosion, but does not cause death of the pupae for a long period of time. In effect the treatment packaged the pupae, prevented eclosion of the fly, and allowed parasites to find pupae over a much longer period of time. This allowed an increase in parasitism at the same time as fewer pupae were produced. This type of control may be helpful in producing larger field populations of parasites, while reducing overall fly populations.

A second trial was conducted with horn flies, with treatment of manure accomplished through the use of a feed additive of methoprene. An increase in parasitism was also noted in horn fly JH trials (Greer 1974).

Natural horn fly parasitism in these trials ranged from 16-34%, as a mixture of hymenopterous pupal parasites. These parasitism rates were increased from 26 to 69% parasitization or 2.6X, when 12 µg/kg methoprene feed additive was used (table 6).

Our studies on horn flies and stable flies, as well as limited trials with house flies, indicate the use of a JH compound which increases parasitism should be considered along with release of parasite species which are demonstrated to be the most important species for each area. The combination allows for increased parasite rates (up to 2.6X), reduced pupal numbers, and much higher F_1 populations of parasites.

Table 5.--The effect of methoprene at 10 ppm EC or SR on stable fly pupal parasitism by naturally occurring populations of *Spalangia nigra*. Four replications were placed in wood chip/paper mill extract media and exposed to field populations of parasites.

Treatment	Total Number Eggs	Pupae Recovered	Pupae Eclosed	% Eclosed	Dead Pupae	% Mortality	Number Parasitized	% Parasitized
Untreated	10,800	3,189	3,049	96%	101	3%	39	1%
ZR515 EC	10,800	1,222	538	44%	512	42%	172	14%
ZR515 SR	10,800	1,034	47	5%	805	78%	182	18%

Table 6.--The effect of methoprene on parasitism rates of the horn fly, *Haematobia irritans*, by naturally occurring populations of pupal parasites. Treatment and check cow pats were replicated and compared for each treatment. Rates are µg/kg/animal.

Replications	Rate	Average # Pupae Recovered	% Parasitism	Treatment Rate	Average # Pupae Recovered	% Parasitism	Increase In Parasitism
7	0	13.4	25.3	24 g/kg	11.9	49.3	1.95X ++
12	0	18.6	26.2	12 g/kg	7.3	68.7	2.6 X ++
8	0	9.6	33.6	6 g/kg	4.9	58.9	1.73X NS
10	0	7.8	16.9	3 g/kg	3.2	50.1	2.96X NS

NS = No significant differences noted between treated and untreated group
++ = Highly significant differences noted between treated and untreated group

HOUSE FLY PARASITISM

Natural parasite rates for the house fly (*Musca domestica*) were determined in poultry houses in north Florida using a pupal trap method.

The pupal collection technique is of significant importance, because it permits sampling of pupae of known age. To date, the only methods of collecting pupae from poultry manure have involved removal of manure samples and flotation of pupae (Legner 1967) or handpicking pupae from the manure (Legner and Bryden 1966, Mitchell et al. 1974). Parasites have been collected by placing emergence traps directly over the manure (Ables and Shepard 1974) or by removing the manure from the farms and placing it in specially designed emergence traps (Legner and Bryden 1966). With pupae of known age, a more accurate determination of the parasite life cycle in the field can be made, as well as percent parasitism determined. Also, the possibility of parasites emerging from diapausing pupae of undetermined age is eliminated. This is important because the diapausing pupae has a greater probability of being parasitized, due to a longer exposure period, and would also give higher false numbers of flies present in the manure.

In the present study, containers of material into which larvae migrated and pupated were utilized to isolate pupae of a known age, so that parasitism rates for a given period of time could be determined. This is important, as parasites in pupae develop over a longer period of time than the normal fly pupal period.

Pupae of known age were obtained from the chicken manure by using cylindrical traps (10 x 20 cm) constructed of 6 mm hardware cloth. These traps were filled with wood shavings and placed in a hole cut into the manure drop area under caged layer hens. Age of pupae was established by leaving the traps in place a specific time period at each site. A biased random sample was taken, since traps were placed in areas where house fly larvae were active. A hole was cut horizontally into the manure cone approximately 5-10 cm above the sand base (Hogsette 1980).

Traps were collected at five or seven day intervals and pupae separated from the wood shavings using water flotation. Pupae were air dried and placed in petri dishes with filter paper discs in the bottom to absorb moisture. The pupae were incubated at 30 ± 1°C and 28 ± 2% RH until parasite emergence was complete. Seven days approximates the time required for emergence of house flies. Initial traps were left in place for five days. This time period was extended to seven days without too great a loss of emerged flies. The number of house fly pupae collected from each trap ranged from 3-170. These

traps seemed specific for house flies, as only 1% of the pupae collected were other than house flies.

The only problem encountered using this technique was the variability in the number of pupae collected. However, by increasing the number of samples per farm, the effect of this variability can be reduced. In most samples, the wood shavings absorbed enough moisture from the surrounding manure, so that they did not float when removing the pupae by flotation.

The percentage of house fly pupae prevented from eclosion ranged from 0 to 42.4% (tables 7-9). Seven day trap periods were chosen as giving the highest parasite rates without excessive loss of flies emerging (tables 8-9).

Although there was a pupal parasite rate of up to 42%, this does not necessarily indicate a decrease in the adult fly population. The ability of flies to migrate from one area to another, especially with open housing, maintains the population over a given area.

Species of parasites recovered from the poultry houses were identified by Dr. Eric Grizelle. Ten pupal parasite species were collected in this trial from house fly pupae (table 10). The most commonly collected were *Spalangia endius*, *S. nigroaenea*, and *S. cameroni*. *Spalangia cameroni* and *S. nigroaenea* had the highest rates of pupal parasitism at 38 and 36%, respectively, followed by *S. endius* at 26%. These data indicate these three parasites would be the most beneficial under north Florida conditions and agree with species identified by Mitchell et al. (1974). The other species were seldom recovered.

Table 7.--Percentage of *Musca domestica* pupae, 1-5 days old, collected 16-21 October 1974, prevented from eclosion by hymenopterous parasites. Using the pupal trap method.

Farm	Total Pupae Collected	Percent Parasitism	Hymenopterous Parasites Emerged	Other Parasites Emerged	Number Unemerged from Unknown Causes
Whitehead	94	5.3	5	0	33
Saunders	237	0.4	1	0	30
Santa Fe	100	0	0	0	17
Pete	52	0	0	0	18
Ayers	259	6.6	17	1	68

Table 8.--Percentage of *Musca domestica* pupae, 1-7 days old, collected 21-28 October 1974, prevented from eclosion by hymenopterous parasites. Using the pupal trap method.

Farm	Total Pupae Collected	Percent Parasitism	Hymenopterous Parasites Emerged	Other Parasites Emerged	Number Unemerged from Unknown Causes
Whitehead	276	42.4	117	4	75
Saunders	107	19.6	21	0	21
Santa Fe	253	17.8	45	0	16
Pete	186	0	0	0	22

Table 9.--Percentage of *Musca domestica* pupae, 1-7 days old, collected 28 October through 4 November 1974, prevented from eclosion by hymenopterous parasites. Using the pupal trap method.

Farm	Total Pupae Collected	Percent Parasitism	Hymenopterous Parasites Emerged	Other Parasites Emerged	Number Unemerged from Unknown Causes
Whitehead	230	5.2	12	0	97
Saunders	331	23.6	78	0	123
Santa Fe	180	28.9	52	0	67
Pete	202	31.2	63	0	65

Table 10.--Species of house fly pupal parasites collected from poultry houses in North Florida using the pupal trap method. Identifications were made by Dr. Eric Grizelle, U. S. National Museum of Natural History, Washington, D.C.

Species Collected	% Total Parasites Collected
1. *Spalangia endius* Walker *	26
2. *S. nigroaenea* Curtis *	36
3. *S. cameroni* Perkins *	38
4. *S. nigra* Latreille	< 1
5. *Muscidifurax raptorellus* Kogan and Legner **	< 1
6. *M. raptor* Girault and Saunders **	< 1
7. *Tachinaephagus zealandieus* Ashmead **	< 1
8. *Pachycrepoideus vindemmiae* (Rondani) **	< 1
9. Staphylinidae *Maseochara* sp. **	< 1

* Most commonly found species
** Present in only one sample

REFERENCES

Ables, J. R., and Shepard M.
1974. Responses and competition of the parasitoids *Spalangia endius* and *Muscidifurax raptor* (Hymenoptera: Pteromalidae) at different densities of house fly pupae. Can. Entomol. 106(8):825-830.

Bishopp, F. C.
1913. The stable fly (*Stomoxys calcitrans* L.), an important livestock pest. Journ. Econ. Entomol. 6:112-126.

Boucek, Z.
1963. A taxonomic study in the *Spalangia* Latreille. Acta Entomol. Mus. Natl. Pragae. 34:429-512.

Butler, J. F.
1973. Unpublished research report.

Combs, R. L., and Hoelscher, C. E.
1969. Hymenopterous pupal parasitoids found associated with the horn fly in Northeast Mississippi. Journ. Econ. Entomol. 62:1234-1235.

Depner, K. R.
1968. Hymenopterous parasites of the horn fly, *Haematobia irritans* (Diptera:Muscidae), in Alberta. Can. Entomol. 100(10):1057-1060.

Escher, R. L.
1977. Pupal parasites of the horn fly *Haematobia irritans* (L.) in North Central Florida. 88 pp. M.S. thesis, University of Florida, Gainesville.
Greer, N. I.
1974. Effect of insect growth regulators on the bionomics and control of the horn fly *Haematobia irritans*. 104 pp. Ph.D. dissertation, University of Florida, Gainesville.
Hogsette, J. A.
1980. The evaluation of poultry pest management techniques in Florida poultry houses. 308 pp. Ph.D. dissertation. University of Florida, Gainesville.
Kramer, R.
1975. Unpublished research report.
Legner, E. F.
1967. Behavior changes the reproduction of *Spalangia cameroni, S. endius, Muscidifurax raptor,* and *Nasonia vitripennis* at increasing host fly densities. Ann. Entomol. Soc. Amer. 60:819-826.
Legner, E. F., and Bryden, H. W.
1966. Suppression of dung inhabiting fly populations by pupal parasites. Ann. Entomol. Soc. Amer. 59:638-651.
Lindquist, A. W.
1936. Parasites of the horn fly and other flies breeding in dung. Journ. Econ. Entomol. 29:1154-1158.
Mitchell, E. R.; Copeland, W. W.; and Tingle, F. C.
1974. Parasites of filth breeding Diptera in poultry houses in North Central Florida. Fla. Entomol. 57:383-384.
Peck, O.
1974. Chalcidoid (Hymenoptera) parasites of the horn fly, *Haematobia irritans* (Diptera:Muscidae), in Alberta and elsewhere in Canada. Can. Entomol. 106:473-477.
Thomas, G. D., and Morgan, C. E.
1972. Parasites of the horn fly in Missouri. Journ. Econ. Entomol. 65:169-174.
Wilkerson, G. G.
1974. A population model for the horn fly *Haematobia irritans* (L.) in North Florida. 109 pp. M.S. thesis, University of Florida, Gainesville.
Wolcott, G. N.
1948. The insects of Puerto Rico: Hymenoptera. Journ. Agric. Univ. Puerto Rico 32:749-873.

LIFE CYCLE: DEVELOPMENT TIMES OF VARIOUS PUPAL PARASITES OF HOUSE FLIES AND HORN FLIES IN FLORIDA

J. F. Butler and R. L. Escher

University of Florida

The length of time required for parasites to complete their life cycles under near natural conditions is important in planning IPM systems. This is a review of field and laboratory studies to develop this information.

MATERIALS AND METHODS

In studies set up to determine pupal parasite levels in the house fly *Musca domestica* (L.) and the horn fly *Haematobia irritans* L., collections of parasitized pupae were made and returned to the laboratory and held for emergence of the parasites. These studies include works by Greer (1974), Wilkerson (1974), Escher (1977), Kramer (1975), and Hogsette (1980).

The collection of house fly pupae was through the use of the pupal trap method (Hogsette 1980). This method allows for pupal age to be limited to a specific time period. In these trials, pupal traps were exposed to house fly larvae migrating into the traps for five to seven days. These traps were then removed to the laboratory. The pupae were separated by flotation and placed in individual containers in an incubator at 30°±1°C and 28±2% R.H. until parasite emergence was complete.

Horn fly pupae were collected from seeded or naturally occurring field populations in artificial or natural cow pats (Escher 1977). These were exposed to parasitism for approximately the time required for the horn fly to reach eclosion. This time period was based on a horn fly developmental formula established by Wilkerson (1974) and varied throughout the year. Horn fly parasitism and developmental data were collected throughout the year and established seasonal developmental rates for the species recovered.

RESULTS

The four most common pupal parasites of the house fly recovered in Northern Florida poultry houses included *Spalangia cameroni* Perkins, *S. nigroaenea* Curtis, *S. endius* Walker, and *S. nigra* Latreille. Other species occurred, but were at rates of less than 1%. The species found were similar to those reported by Mitchell et al. (1974). Identification of parasites was verified by E. Grizelle, U.S. National Museum.

Pupal Parasite Developmental Rates for House Flies

The percent parasitism and the mean time required for development of house fly pupal parasites are listed in table 1. These developmental rates are at the exposed field temperature for seven days and then 30°C until emergence. The high temperature in the test chamber was utilized to approximate manure temperature due to bacterial action. Of the parasites recovered from the five poultry farms, *S. cameroni* was most prevalent at 38%, followed by *S. nigroaenea* at 36%, then *S. endius* at 26%. *S. nigra* was recovered only once in this survey.

Mean days for development was the shortest for *S. nigra* at 16 days from a single male specimen. *S. endius* required 17.7, while *S. nigroaenea* required 21.9 and *S. cameroni* required 24.

The developmental rates for pupal parasites observed in this study are at least twice that of the house fly when reared at the same temperature. Some species take three times as long to develop. This makes it difficult for parasite numbers to build up to effectively limit house fly populations. In a parallel study to this research, a parasite release program was conducted, but did not significantly change parasite rates from the naturally occurring rates (Kramer 1975). Others (Morgan et al. 1975, Weidhaas et al. 1977) have demonstrated that only high rates of parasite release on a biweekly time period were effective in controlling house flies. The sex ratio of reared parasites was approximately the same for *Spalangia endius* at ♀:♂ 1:0.4 and *S. nigroaenea* at ♀:♂ 1:0.3. *S. cameroni* had a sex ratio of ♀:♂ 1:0.8, when season of the year was ignored.

Pupal Parasite Developmental Rates for Horn Flies

Horn fly parasites belonged to the same species as those found in house fly pupae with the exception of *S. haematobiae* which seems to be species specific.

Seasonal collections for species of horn fly pupal parasites are given in table 2 for *S. cameroni*, table 3 for *S. haematobiae*, table 4 for *S. nigroaenea*, table 5 for *S. nigra*, and table 6 for *S. endius*. Sex ratios for *S. cameroni* (table 2) were approximately 1 ♀ and 0.8 ♂ in the early season, from April through September, then showed a decline in ♂ in the

late fall, from October through December. *S. haematobiae*, *S. nigroaenea*, and *S. nigra* (table 3) had much lower numbers of males, but the ratios were variable.

Comparison of the mean days for development for the *Spalangia* sp. are shown when seasonal considerations are made (figure 1). The most common and probably the most important horn fly parasites were those with intermediate developmental rates; these were *S. cameroni* and *S. haematobiae*. Even during the warmest months, the developmental rates required at least 21 days, which approximates three times the time required for horn fly development.

REFERENCES

Escher, R. L.
1977. Pupal parasites of the horn fly *Haematobia irritans* (L.) in North Central Florida. 88 pp. M.S. thesis, University of Florida, Gainesville.

Greer, N. I.
1974. Effects of insect growth regulators on the bionomics and control of the horn fly *Haematobia irritans*. 104 pp. Ph.D. dissertation, University of Florida, Gainesville.

Hogsette, J. A.
1980. The evaluation of poultry pest management techniques in Florida poultry houses. 308 pp. Ph.D. dissertation, University of Florida, Gainesville.

Kramer, R.
1975. Unpublished research report.

Mitchell, E. R.; Copeland, W. W.; and Tingle, F. C.
1974. Parasites of filth-breeding Diptera in poultry houses in North Central Florida. Fla. Entomol. 57:383-384.

Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Weidhaas, D. E.; and Benton, A.
1975. Suppression of a field population of house flies with *Spalangia endius*. Science 189:388-389.

Weidhaas, D. E.; Haile, D. G.; Morgan, P. B.; and LaBrecque, G. C.
1977. A model to simulate control of house flies with a pupal parasite, *Spalangia endius*. Environ. Entomol. 6(4): 489-500.

Wilkerson, G. G.
1974. A population model for the horn fly *Haematobia irritans* (L.) in North Central Florida. 109 pp. M.S. thesis, University of Florida, Gainesville.

Table 1.--Summary of *Spalangia* parasitism and developmental rates from house fly pupae from North Florida poultry houses using the pupal trap method. Time period October through November.

Species	Total Collected	♀	♂	Sex Ratio ♀:♂	Rate of Parasitism	Days for Mean Development	Standard Error of Mean	Standard Deviation
S. cameroni	297	168	129	1:0.8	38%	24.0	1.84	4.12
S. endius	206	152	54	1:0.4	26%	17.7	0.9	2.03
S. nigroaenea	282	217	65	1:0.3	36%	21.9	2.3	5.02
S. nigra	1	0	1	---	---	16.0	---	---

Table 2.--*Spalangia cameroni* parasitism and developmental rates in horn fly pupae from larval seeded artificial cow pats.

Month	Total	♀	♂	Ratio ♀:♂	Parasitism Rate/Month	Days for Mean Development	Standard Error of the Mean	Standard Deviation
Apr	5	3	2	1:0.7	0.4	32	1.57	3.51
May	65	40	25	1:0.6	5.4	26	0.28	2.25
June	197	103	94	1:0.9	16.5	24	0.12	1.76
July	120	72	48	1:0.7	10.0	25	0.18	1.94
Aug	124	64	60	1:0.9	10.4	25	0.20	2.27
Sept	55	34	21	1:0.6	4.6	26	0.33	2.47
Oct	6	5	1	1:0.2	0.5	27	0.56	1.37
Nov	29	22	7	1:0.3	2.3	34	0.94	4.92
Dec	10	5	5	1:1	0.8	35	0.61	1.91
Jan	---	---	--	---	---	--	---	---
Feb	8	8	0	---	0.7	35	2.10	5.95

Table 3.--*Spalangia haematobiae* parasitism and developmental rates in horn fly pupae from larval seeded artificial cow pats.

Month	Total	♀	♂	Ratio ♀:♂	Parasitism Rate/Month	Days for Mean Development	Standard Error of the Mean	Standard Deviation
May	4	4	0	---	0.3	29	3.0	5.20
June	9	7	2	1:0.3	0.8	23	0.8	2.50
July	37	28	9	1:0.3	3.1	24	0.4	2.48
Aug	55	49	6	1:0.1	4.6	23	0.2	1.61
Sept	35	30	5	1:0.2	2.9	24	0.5	2.95
Oct	9	9	0	---	0.8	24	0.6	1.76
Nov	33	27	6	1:0.2	2.8	32	0.5	3.20
Dec	7	5	2	1:0.4	0.6	32	0.1	0.38
Jan	--	--	--		---		---	---
Feb	78	45	33	1:0.7	6.5	37	7.0	6.06

Table 4.--*Spalangia nigroaenea* parasitism and developmental rates in horn fly pupae from larval seeded artificial cow pats.

Month	Total	♀	♂	Ratio ♀:♂	Parasitism Rate/Month	Days for Mean Development	Standard Error of the Mean	Standard Deviation
May	3	2	1	1:0.5	0.3	25	2.0	3.46
June	9	8	1	1:0.1	0.8	22	0.5	1.41
July	7	4	3	1:0.8	0.6	21	0.5	1.34
Aug	41	20	21	1:1	3.4	22	0.4	2.52
Sept	11	9	2	1:0.2	0.9	23	0.4	1.18
Oct								
Nov	26	20	6	1:0.3	2.2	26	0.2	0.79
Dec								
Jan								
Feb	31	5	8	1:1.6	1.1	31	0.2	0.83

Table 5.--*Spalangia nigra* parasitism and developmental rates in horn fly pupae from larval seeded artificial cow pats.

Month	Total	♀	♂	Ratio ♀:♂	Parasitism Rate/Month	Days for Mean Development	Standard Error of the Mean	Standard Deviation
May	14	8	6	1:0.8	1.2	30	0.7	2.7
June	80	61	19	1:0.3	6.7	28	0.1	1.1
July	25	24	1	1:0.04	2.1	27	0.6	3.2
Aug	1	1	0	---	0.1	24	0.0	0.0
Sept	3	1	2	1:2	0.3	30	3.0	5.2
Oct								
Nov								
Dec	3	3	0	---	0.3	43	0.0	0.0
Jan	4	3	1	1:0.3	0.3	59	1.1	2.2
Feb	9	5	4	1:0.8	0.8	51	0.8	2.2

Table 6.--*Spalangia endius* parasitism and developmental rates in horn fly pupae from larval seeded artificial cow pats.

Month	Total	♀	♂	Ratio ♀:♂	Parasitism Rate/Month	Days for Mean Development	Standard Error of the Mean	Standard Deviation
May	3	1	2	1:2	0.3	29	0.9	1.5
June	56	45	11		4.7	21	0.2	1.6
Nov	1	0	1	---	0.1	27	---	---

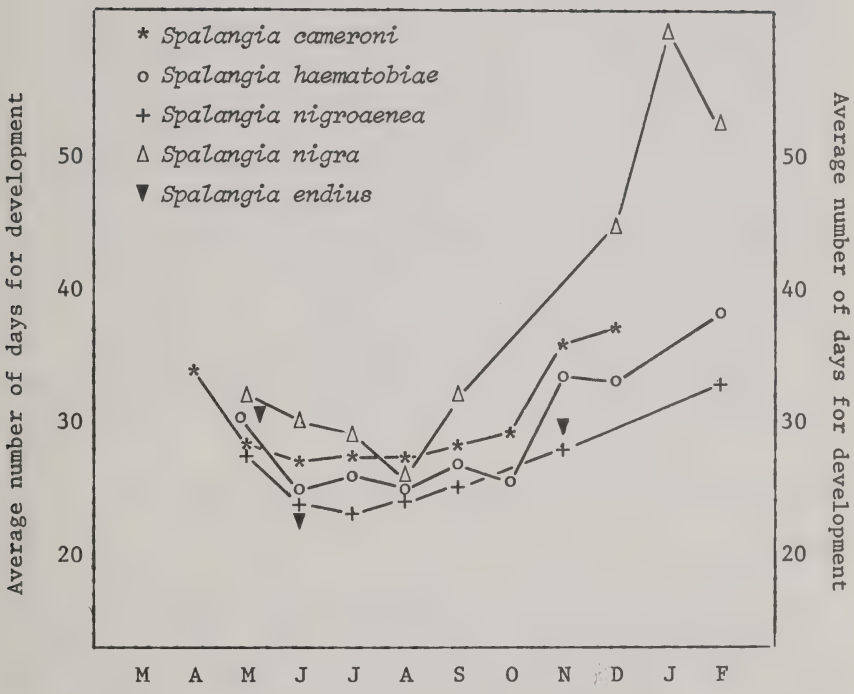


Figure 1.--Comparison of seasonal development times for the four major and one minor (*S. endius*) hymenopterous pupal parasites of the horn fly.

FIELD AND LABORATORY DEVICES FOR MANIPULATION OF PUPAL PARASITES

J. A. Hogsette and J. F. Butler

U.S. Department of Agriculture and University of Florida

INTRODUCTION

The need to evaluate field populations of pupal parasites has become increasingly important with the advent of Integrated Pest Management (IPM) programs. The devices described below can be used for surveying parasite populations using laboratory-reared or wild fly pupae, determining the number of parasites that emerge from each pupal case, and for collecting wild pupae of known age for determination of a rate of parasitism.

TISSUE CULTURE PLATES FOR HOLDING PARASITIZED PUPAE

Description

Tissue culture plates are a grouping of small clear plastic tubes or chambers fastened together to form a small tray with a tight-fitting lid. The tissue culture plate most useful for our work was 85 x 125 x 15 mm with 96 chambers (figure 1).

Application

Tissue culture plates are convenient for holding fly pupae which are suspected of being parasitized. Each plate accomodates a sample size of 96 or less. One pupa is placed in each chamber and the lid is secured with rubber bands. Since the plates are clear, each pupa in the group can be viewed individually. As parasites eclose, symbols denoting eclosure date can be made on the lid over the appropriate cells. Besides eclosure date, tissue culture plates can be used to determine the rate of parasitism and to determine the number of parasites eclosing from each pupa.

Figure 1.--Tissue culture plate with fly pupae.

PUPAL TRAPS--A METHOD FOR COLLECTING FLY PUPAE OF KNOWN AGE

Description

These traps are cylinders 30 cm high by 10 cm in diameter with sides and bottoms made of hardware screen (figure 2). When loosely filled with moist wood shavings and set in areas where fly larvae are active, they become attractive pupation sites. These traps are set in holes made in fly breeding media with a golf course plugger. The diameter of the trap should correspond to the size of the plugger used to set the traps. Holes should be made in a nearly horizontal position just below (2 cm) the surface of the fly breeding medium. After traps have been inserted into medium it should be lightly tamped around the traps and the locations flagged so traps can be relocated with ease.

Field Application

The significant aspect of pupal traps is that they can be used to collect fly pupae of known age. This is advantageous when determining rates of parasitism from naturally-occurring or mass-released hymenopteran pupal parasites and when field-testing compounds such as IGR's which affect flies in the pupal stage.

Traps should be left in the field for at least 7 days to minimize variation between traps. Pupae are separated from wood

shavings by immersing the trap contents in water for 15 to 30 min. Pupae can be skimmed from the water surface, dried and placed in covered petri dishes for 24 to 72 hrs. to allow flies to eclose. Noneclosed pupae can then be put into tissue culture plates to await parasite eclosure.

Figure 2.--Tagged pupal trap after removal from manure pack.

TRAPS FOR SURVEYING FIELD POPULATIONS OF PARASITES OF FLIES

Description

These parasite traps are made from 20-dram clear plastic vials with twist off lids. The entire bottom and a portion of the lid are cut out and replaced with window screen. Traps are baited with newly formed fly pupae and placed in the field near fly breeding and/or pupation sites. These baited traps are highly attractive to the hymenopteran parasites as well as other parasites. Parasites have been observed entering traps and stinging pupae less than 5 min. after traps were placed in the field.

Modifications

If traps are placed in relatively dry areas it may be appropriate to place them directly on the ground. However, if trap placement areas are wet and there is a possibility of liquid entering the traps, traps can be fastened on standard metal tent stakes with rubber bands and suspended above the placement area (figure 3). Care must be taken to keep the traps as close to the ground as possible so they can be easily

detected and reached by the parasites. Besides being attractive to the parasites, parasite traps are also attractive to ants. Pupae in baited traps can be completely decimated by ants in 24 hrs. Attacks by ants can be prevented by fastening the traps to the stakes and painting a ring of adhesive (Tack Trap) on the stake above the soil line. Stakes also prevent traps from being moved from their original location by birds or other animals.

Figure 3.--Parasite trap on stake with foam sleeve.

Baited traps cannot be placed in sunny areas unless they are protected from the sun. Removable sleeves fashioned from 25 mm foam rubber will insulate the traps in sunny locations and reduce mortality from the heat by 50%. Sleeves are slipped onto the traps before the traps are fastened to the stakes (figure 3).

Field Application

Pupal traps are good tools for surveying native populations of parasites. Traps are usually left in the field for a maximum of three days. Tests have shown that attempts by some species of wasps to parasitize house or stable fly pupae greater than two days old were largely unsuccessful (Morgan et al. 1975, 1978, 1979). Also, extended periods in the field increase the chances for superparasitism to occur.

After traps are collected from the field, pupae are kept in the traps until flies from the unparasitized pupae eclose.

Noneclosed pupae are then transferred to tissue culture plates and held 60 days for parasite eclosure.

REFERENCES

Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Weidhaas, D. E.; Benton, A.; and Whitfield, T.
1975. Rearing and release of the house fly pupal parasite Spalangia endius Walker. Environ. Entomol. 4: 609-611.

Morgan, P. B.; Weidhaas, D. E.; and LaBrecque, G. C.
1978. Host-parasite relationship of the house fly, Musca domestica L. and the microhymenopteran parasite, Pachycrepoideus vindemiae (Rondani). Southwestern Entomol. 3: 176-181.
1979. Host-parasite relationship of the house fly, Musca domestica L. and the microhymenopteran pupal parasite, Muscidifurax raptor Girault and Sanders (Diptera: Muscidae and Hymenoptera: Pteromalidae). J. Kans. Entomol. Soc. 52: 276-281.

IMPORTANCE OF MONITORING HOUSE FLY AND STABLE FLY IMMATURE AND ADULT POPULATIONS IN IPM PROGRAMS USING BIOCONTROL

R. S. Patterson

U.S. Department of Agriculture

At this workshop, we have heard about the successes and failures of biocontrol in integrated pest management programs to suppress flies at poultry and livestock complexes. Biocontrol can be integrated into any IPM fly control program and be very beneficial if one understands it and uses it properly. Biocontrol alone, specifically the use of parasites and/or predators, is a slow and expensive way to control flies. Often the effectiveness of these organisms is overstated in both sales literature and research grant proposals capitalizing on the public's fear of pesticides contaminating the environment.

There are two types of approaches for the release of parasites and predators into the environment to control nuisance host populations such as house flies and stable flies. The classical approach is when the parasites and/or predators are introduced or inoculated into the environment through one or more releases. The second approach is to inundate by sustained massive releases of laboratory reared parasites and/or predators into the environment to overwhelm the host populations. There are certain merits to each approach. In the inoculative releases the objective is usually to reestablish native organisms which were eradicated due to poor farm management and/or excessive use of pesticides. Sometimes it involves the establishment of an exotic species, which hopefully may outcompete the native ones and suppress the host below an economic threshhold. Usually it takes months, sometimes years, for the parasites and/or predators to build up to a level in the environment, which causes a noticeable effect on a fly population. Many times the new parasites or predators cannot compete with other natural enemies, or climatic conditions may be adverse and the organisms die out prior to establishment. Yet, inoculative releases are relatively inexpensive based on an overall cost-benefit ratio. Sustained or inundative releases, such as those carried out by Morgan et al. (1975, 1976b) and most

commercial applicators, give usually a quick response with a high degree of fly suppression similar to pesticides or sterile male releases. However, it does have a low cost-benefit ratio if carried out for a long period. According to Weidhaas (1981), sustained releases if timed and applied correctly can give seasonal fly control at a cost-benefit ratio equal to or better than coventional pesticide programs. The reason is that most parasites and predators of flies have a very low biotic potential in comparison to that of house flies or stable flies. Consequently, natural parasitism affects only about 2% of the fly pupae in the spring but normally the number of parasitized pupae increases as the fly season progresses (Stage and Petersen 1981). If sustained releases are undertaken in the spring using a proven native or exotic parasite species, they can complement the natural fauna to control wild fly populations. The wild fly population will be suppressed and the subsequent F_1 progeny of these released insects will be present to suppress future pest populations during the remaining fly season. The parasite releases have to be of sufficient duration to cover the life cycle of the parasites or predators being released, which may be a month or more. These sustained releases work basically the same as the sterile male technique where a known ratio of host to parasite is set up in order to obtain a desired degree of suppression based on the competitiveness of the released insect (Knipling 1979). Sustained releases do have the bonus effect of the subsequent progeny from the released organisms which the sterile insects do not have. However, one must know the size and species complex of the host population, its ecology and biology. Also one must be knowledgeable about the parasites and/or predators being released and their competitiveness in the wild as well as their efficiency in attacking the target host population. The efficiency of various parasites has been reported by various researchers (Ables and Shephard 1974a,b, 1976; Butler et al. 1981; Legner and Gerling 1967; Legner and Dietrick 1972; McCoy 1965; Morgan et al. 1976a, b; Morgan 1981). This research was done both in the laboratory and in the field. The parasites were released and fresh pupae were collected and checked for parasitism and the degree of fly suppression was recorded. Morgan et al. (1976) determined the degree of parasitism by collecting wild pupae from the release site. This has the disadvantage that uneclosed parasitized pupae remain in the field for 19-30 days or longer, whereas non-parasitized pupae eclose within 3-5 days. This causes a bias in favor of the parasites; however, it can be adjusted if the time of parasite eclosion is recorded. Finding host pupae in manure is sometimes difficult especially if the host population is low. Axtell (1981) and Hogsette and Butler (1981) prefer to use sentinel host pupae and/or traps filled with favorable pupation media to trap newly-formed pupae. These techniques have the advantage that one knows the age of the pupae and the daily rate of parasitism. By flagging these sentinel pupae and pupae traps there is no problem in locating host material even when fly populations are low.

Morgan et al. (1976) found that the hymenopteran parasite *S. endius* was not as attracted to sentinel pupae as wild ones in the field. It has been theorized that the repeated washing of the pupae during the mass rearing may have reduced a natural pheromone on the surface of the pupae which is used by the parasite to detect the host in the field. Regardless of the technique used to evaluate the organism being released, one should know if the organism is attacking the host. Recent studies by Stage and Petersen (1981) found that after releasing over 2 million *Nasonia vitripennis* Walker and surveying 12,000 wild fly pupae only one was parasitized by this parasite. Likewise, Hogsette (1981) in his study found that the parasites were being released where adult flies were but the immature breeding was taking place almost a kilometer away. All the manure from the stable area was being carried to a dump a km away daily, yet most of the parasites were left behind to be killed by the overhead pesticide mists. At the dump less than 1 percent of fly pupae sampled were parasitized by the wasps being released. The cost of this parasite release program at this horse complex was $800/month. In both cases if the biology and ecology of the host and parasite were understood these failures could have been avoided. Many times successes are claimed when the decline in fly population was due to natural climatic conditions or changes in fly-breeding habitats. The release of parasites or predators have had little effect. Often the suspension of larvicides has the biggest effect, as numerous natural parasites and predators reestablish themselves and set up a natural biological balance in the environment as noted by Dietrick (1981). The natural and artificial changes which affect the fly population seasonally can only be verified if they are properly monitored.

The immature or larval stages of either the house fly or stable fly can be found by searching the suitable breeding media on the premises or surrounding area. Adult flies can migrate long distances from their breeding sites. Stable flies have been recorded to move as much as 117 km (Rogers 1968). Distances of several km are common for both fly species. Therefore, release of parasites at sites where adult flies are a problem often is uneffective. If most of the larval breeding sites are known, then one can easily estimate the size of the immature fly population and establish life history models which estimate the number of parasites needed to suppress the adult fly population (Weidhaas 1981).

Adult house fly populations can be monitored using grids, traps, sticky tapes and spot cards. Each technique has its own merits and individual scientists prefer certain techniques over another. Morgan et al. (1981) uses the modified Scudder Grid (Murvosh and Thaggard 1966) and finds it very satisfactory. It has the advantage in that the results are known immediately. The grid is normally placed where the flies congregate and counts are made for a set time. As long as

care is taken to maintain consistency in time of day, counting time and location each time the grid is used it is a very reliable method. The biggest drawback is the variance between individuals using the grid. Sticky traps or ribbon are widely used to assay fly populations inside barns, sheds, etc. They work well if placed in strategic locations where the flies will rest on them. A modification of this technique was developed by Patterson et al. (1980) where inch-wide and 18 inch-long fiberglass strips were coated with "Tack Trap" and hung similar to fly tapes. These strips are rigid and can be taken back to the laboratory and the flies removed for identification. The strips can be easily cleaned, retreated and reused repeatedly throughout the season. The Williams' trap used for stable flies (Williams 1973) will also catch large numbers of house flies, which use it as a resting site, around manure dumps, silage pits and garbage pits. All sticky traps have to be changed after a few hours or days otherwise they become overcrowded with the corpses of dead flies and other insects caught in the adhesive. Dust can also be a problem on sticky traps unless they are changed regularly.

Baited traps and jugs have been used very successfully by Carlson (1973) and Rutz and Axtell (1979). A toxic bait is placed in the container and the flies feeding on the material die and remain in the tray or jug. Placement of the containers is critical as they have to be accessable to the flies but out of reach of children and livestock which might be attracted to the toxic bait. The baited jugs work best when suspended by a wire over or near the fly breeding media. The Dodge Trap or a modification of it works very well for house flies if baited with an aged mixture of yeast, ammonium carbonate and water, or the bait described by Pickens et al. (1973). If placed outside, these traps will collect large numbers of house flies in a few hours. However, if they are not cleaned every 24-48 hrs the dead flies will attract mainly blow flies and become ineffective for house flies.

The spot cards described by Axtell (1970) work fine for monitoring populations if placed where flies rest. The number of fly specks gives an indication of the number of flies in the area. Again, these have to be changed often otherwise it becomes difficult to count.

To monitor stable fly populations two techniques have been found to be very satisfactory for field studies. The sticky trap as modified by Williams (1973) is the best. Certain types of clear, flat fiberglass (Alsynite and Filon are two brands of fiberglass that work for stable fly traps) reflect ultraviolet light in a range that is very attractive to stable flies. These traps, if placed 30-90 cm above the ground will attract any stable flies that are within visual range. These traps are so attractive, that they have been modified and used to control wild stable fly populations (Meifert et al 1978).

Since attraction relies upon a visual response, the traps must be placed where the flies can see them. Also, of importance is the adhesive used over the fiberglass, as some will mask the reflectancy and reduce the attractancy of the trap. The best adhesive for the stable fly traps is polyisobutylene (this is the basic chemical in the commercial product "Tack Trap"). Some of the adhesives which are commonly sold can reduce the efficiency of the traps as much as 80-90%. Yet, superficially these sticky materials appear the same. The Williams' trap works best in the direct sunlight but can also be used in shaded areas. It does not work well inside buildings. Placement of traps influences catch, and in heavy fly populations the traps have to be changed often as the captured flies and other insects block the reflected light and reduce the attractancy of the trap. Dust and dirt are also a problem with this trap if placed in areas where animals create a lot of dust, such as riding rings, feedlots, etc.

The other method for monitoring stable flies was described by LaBrecque et al. (1975) where fly counts are made on the animals. Stable flies normally go to the animal especially the legs and hocks, and stay long enough to take a blood meal. The flies take at least one blood meal a day. It has been estimated that for every fly observed feeding on an animal there are approximately 55 resting away from the animal. Based on this observation, an estimate of the size of a wild stable fly population in the area can be made if the number of animals in the area are known and the fly counts from 10 or more animals in each herd. Patterson et al. (1979) on St. Croix was able to establish the stable fly population in a 125 km^2 area. These figures on population size by fly counts on animals were also verified by marked-release Lincoln indexes and larval surveys at immature breeding sites. The fly counts on animals is an easy quick method. The main difficulty is getting close enough to observe the flies and to differentiate between stable flies and other insects on the animal. On St. Croix binoculars were used to see the flies.

All of these monitoring techniques have advantages and disadvantages for surveying house fly and stable fly populations. Yet, they are essential to obtain the necessary data to prove that parasites and/or predators are an essential part of any fly IPM program.

REFERENCES

Ables, J. R., and Shephard, M.
1974a. Hymenopterous parasitoids associated with poultry manure. Environ. Entomol. 3: 884-886.
1974b. Responses and competition of the parasitoids Spalangia endius and Muscidifurax raptor (Hymenoptera: Pteromalidae) at different densities of house fly pupae. Can. Entomol. 106: 825-830.

1976. Seasonal abundance and activity of indigenous Hymenopterous parasitoids attacking the house fly (Diptera: Muscidae). Can. Entomol. 108: 841-844.

Axtell, R. C.
1970. Integrated fly control program for caged-poultry houses. J. Econ. Entomol. 63: 400-405.
1981. Use of predators and parasites in filth fly IPM programs in poultry housing. Proc. of Workshop on Status of Biological Control of Filth Breeding Flies, Gainesville, Florida, February 4-5, 1981.

Butler, J. F.; Escher, R.; and Hogsette, J. A.
1981. Natural parasite levels in house flies, stable flies and horn flies in Florida. Proc. of Workshop on Status of Biological Control of Filth Breeding Flies, Gainesville, Florida, February 4-5, 1981.

Carlson, D. A.
1973. A better attractant for house flies. USDA Agric. Res. 21: 8-9.

Dietrick, E. J.
1981. Commercial production and use of predators and parasites for fly control programs. Proc. of Workshop of Status of Biological Control of Filth Breeding Flies, Gainesville, Florida, February 4-5, 1981.

Hogsette, J. A.
1981. Fly control by composting manure at a South Florida equine facility. Proc. of Workshop on the Status of Biological Control of Filth Breeding Flies, Gainesville, Florida, February 4-5, 1981.

Hogsette, J. A., and Butler, J. F.
1981. Field and laboratory devices for manipulation of pupal parasites. Proc. of Workshop on Biological Control of Filth Breeding Flies, Gainesville, Florida, February 4-5. 1981.

Knipling, E. F.
1979. The basic principles of insect population suppression and management. USDA Agric. Hndbk. 512. 659 pp.

LaBrecque, G. C.; Bailey, D. L.; Meifert, D. W.; and Weidhaas, D. E.
1975. Density estimates and daily mortality rate evaluation of stable fly (Stomoxys calcitrans) (Diptera: Muscidae) populations in field cages. Can. Entomol. 107: 597-600.

Legner, E. F., and Dietrick, E. J.
1972. Innundation with parasitic insects to control filth breeding flies in California. Proc. 40th Annu. Conf. Calif. Mosq. Contr. Assoc., Inc. January 31-February 2. p. 129-130.

Legner, E. F., and Gerling, D.
1967. Host feeding and oviposition on Musca domestica by Spalangia cameroni, Nasonia vitripennis and Muscidifurax raptor (Hymenoptera: Pteromalidae)

influence their longevity and fecundity. Ann. Entomol. Soc. Am. 60: 678-691.

McCoy, C. W.
1965. Biological control studies of Musca domestica and fannia sp. on southern California poultry ranches. Proc. 33rd Annu. Conf. Calif. Mosq. Contr. Assoc., Inc., January. pp. 40-42.

Meifert, D. W.; Patterson, R. S.; Whitfield, T.; LaBrecque, G. C.; and Weidhaas, D. E.
1978. Unique attractant-toxicant system to control stable fly populations. J. Econ. Entomol. 71: 290-292.

Morgan, P. B.
1981. The potential use of parasites to control Musca domestica L. and other filth breeding flies in agricultural installations in the southern United States. Proc. of Workshop on Status of Biological Control of Filth Breeding Flies, Gainesville, Florida, February 4-5, 1981.

Morgan, P. B.; Patterson, R. S.; and LaBrecque, G. C.
1976a. Host-parasitoid relationship of the house fly Musca domestica L., and the protelean parasitoid Spalangia endius Walker. J. Kans. Entomol. Soc. 49: 483-488.
1976b. Controlling house flies at a dairy installation by releasing a protelean parasitoid Spalangia endius Walker. J. Ga. Entomol. Soc. 11: 39-43.

Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Weidhaas, D. E.; Benton, A.; and Whitfield, T.
1975. Rearing and release of the house fly pupal parasite Spalangia endius Walker. Environ. Entomol. 4: 609-611.

Murvosh, C. M., and Thaggard, C. W.
1966. Ecological studies on the house fly. Ann. Entomol. Soc. Am. 59: 533-547.

Patterson, R. S.; LaBrecque, G. C.; and Williams, D. F.
1979. Use of the sterile male technique as an adjunct to insecticidal and physical methods for stable fly control on the island of St. Croix, USVI. Proc. of the Symp. IAEA, Vienna, May 7-11, 1979. pp. 283-295.

Patterson, R. S.; Meifert, D. W.; Buschman, L.; Whitfield, T.; and Fye, R. L.
1980. Control of the house fly Musca domestica (Diptera: Muscidae) with permethrin coated fiberglass strips. J. Med. Entomol. 17: 232-234.

Pickens, L. G.; Miller, R. W.; and Mowry, G. R.
1973. An improved bait for flies (Diptera: Muscidae: Calliphoridae). J. Med. Entomol. 10: 84-88.

Rogers, A.
1971. West Florida arthropod research laboratory (studies of the dispersal of adult dog flies). Annu. Rpt. Div. of Health, Fla. Dept. of HRS. pp. 103-104.

Rutz, D. A., and Axtell, R. C.
1979. Sustained releases of Muscidifurax raptor (Hymenoptera: Pteromalidae) for house fly (Musca domestica) control in two types of caged-layer poultry houses. Environ. Entomol. 8: 1105-1110.

Stage, D. A., and Petersen, J. J.
1981. Mass release of pupal parasites for control of stable flies and house flies in confined feedlots in Nebraska. Proc. of Workshop on Status of Biological Control of Filth Breeding Flies, Gainesville, Florida, February 4-5, 1981.

Weidhaas, D. E.
1981. Computer simulations of the interaction of a pupal parasite during aumentative releases for control of the house fly. Proc. of Workshop on Status of Biological Control of Filth Breeding Flies, Gainesville, Florida, February 4-5, 1981.

Williams, D. F.
1973. Sticky traps for sampling populations of Stomoxys calcitrans. J. Econ. Entomol. 66: 1274-1280.

STICKY PANELS AS TRAPS FOR MUSCA AUTUMNALIS

Lawrence G. Pickens

U.S. Department of Agriculture

In an effort to reduce sample variability and increase sample precision, we used sticky traps as an alternative method of surveying face fly, Musca autumnalis De Geer, populations on pastured cattle in Maryland.

We found that white panels which reflect a large quantity of light in the ultraviolet region of the spectrum were more attractive to face flies than were colored or low-uv. reflecting materials.

The most efficient trap which we tested was a glossy white pyramid (tetrahedron) which was made of four 2 ft wide by 2 ft high triangles joined at 2 edges. The best trap placement was ca. 1 meter above the ground, in full sunlight and within 5 meters of spots where cattle passed by or congregated. Locations in shallow valleys near trees and water were better than locations in open fields or on hills. The panels should not be placed south of the cattle if it can be avoided, as such a placement results in the shaded north trap face being towards the cattle. The traps must be protected from cattle.

The pyramids were covered by clear, transparent plastic and painted with a thin layer of tack trap. The plastic was removed and replaced every 1-2 days for maximum sampling efficiency.

Although the traps captured up to 20% of marked and released flies in 24 hrs in some tests, the average percentage recapture of marked flies on 6 farms over 3 years indicates that it will normally require 20-30 traps to capture 10-20% of the flies per day.

Total catch seems to be more nearly correlated with total traps per farm than with traps/head of cattle, traps/acre, or no. of trapped sites.

As an average, 6 traps were necessary for an estimate with $P>0.90$ and an error of ±2% of the daily catch. The traps correctly distinguished between two marked fly populations which differed in size by 25% 15 times out of 15 trials, compared with correct identifications 6 times by counting the flies on the faces of 15 cattle (Pickens et al. 1977).

REFERENCE

Pickens, L. G.; Miller, R. W.; and Grasela, J. J. 1977. Sticky panels as traps for *Musca autumnalis*. J. Econ. Entomol. 70: 549-552.

FLY CONTROL BY COMPOSTING MANURE AT A SOUTH FLORIDA EQUINE FACILITY

Jerome A. Hogsette

U.S. Department of Agriculture

INTRODUCTION

The horse industry in Florida is a large, rapidly expanding industry whose economic value cannot be measured by the standards established for other types of livestock. The formation of large equine facilities in the central and southern portions of the state has created habitats suitable for excessive house and stable fly breeding. This report describes how fly control was attained at one large facility by the development of an integrated pest management program based on manure management techniques.

THE PROBLEM

The test facility has a capacity for stabling 300 to 400 horses in permanent stalls which readily accomodates the 80 to 100 horses kept at the facility during the summer and early fall. During the early winter and spring season when horse populations average 400 and peak at 1,000 to 1,500 in February, temporary stalls under large tents are used to house the additional animals. The nature of the problem can easily be appreciated when it is noted that working horses produce an average of 7.3 kg of manure and wet stall litter per day (Morrison 1966). Simple multiplication will show that the daily manure and stall litter accumulation at the test facility will average 2,909 kg during the early winter and spring season and 591 to 727 kg during the remainder of the year.

Sanitation at the permanent stables was excellent but very poor at the temporary stables. When manure and stall litter was picked up from stables, it was taken to a manure dump by a truck and either windrowed or dumped in individual piles. This provided maximum surface area for potential fly breeding. The poor drainage at the dump site kept manure and stall litter wet enough to allow flies to breed in large numbers at the soil

interface. Standing pools of manure-enriched water encouraged mosquito breeding. The dump site was situated less than 91 m from one of the permanent stabling areas and ca. 182 m from a restaurant. Several small unscreened food pavilions were located between the dump and the restaurant. Large numbers of house flies swarmed around the restaurant and food pavilions and made comsumption of food in outdoor areas an aggravating experience. Stable flies in stable and paddock areas annoyed horses, riders and spectators. Attempts to interest nurserymen in the manure and stall litter were unsuccessful as were other attempts to develop a satisfactory manure disposal and removal system. Manure at the dump was not properly managed and excessive fly breeding was the result.

THE EXISTING METHOD OF FLY CONTROL

A commercial supplier of hymenopteran pupal parasites had been under contract at the test facility for ca. 5 months. The supplier contacted the facility once a week by phone, determined how many horses were on the premises and thereby decided how many parasites to release during that particular week. Parasites were shipped by United Parcel Service and released by personnel at the test facility. The last two shipments of parasites were released by the supplier or his representative.

Personnel at the test facility were instructed by the parasite supplier to release the parasites by scattering the parasitized fly pupae in the permanent stalls. Parasites would eclose and prevent fly breeding in the stalls. Some parasites would be carried to the manure dump in the stall litter when the stalls were cleaned and thus prevent fly breeding at that location. According to the parasite supplier, the effects of the overhead spray systems operating in each of the permanent stalls would be minimal on the parasites. The last two releases of the study (800,000 *Muscidifurax raptor*, 400,000 per release) were scattered on the ground at the dump. The overall cost of this program was ca. $800 per month.

Four days after the last parasite release, 975 newly-formed fly pupae were recovered from the dump and held in the laboratory for parasite emergence. Six pupae had been parasitized by *M. raptor* and 66 by *Spalangia nigroaenea*, a native species. This constituted parasitism rates of 0.62% and 6.77%, respectively. As a result of these findings, the parasite program was suspended at the suggestion of the supplier.

REASONS FOR COMPOSTING

Since manure removal could not be arranged and a fly control program at the manure dump was lacking, composting seemed to be a feasible alternative. The original consideration was to compact the manure and stall litter into a form which would reduce the surface area and hence reduce fly breeding with

minimal use of pesticides. Secondly, it was hoped that composting could be a means for transforming daily accumulation of waste into a usable product.

PROBLEMS WITH COMPOSTING

One problem with composting other than lack of experience by most farmers is the lack of specialized equipment. This includes sieves, grinders and other machinery needed for manipulating the manure and stall litter before, during and after the composting process. Dump trucks, front-end loaders and bulldozers were available for moving and stacking manure and stall litter but the large volume of material involved and the limited area available as a composting site made stirring or turning the compost an impractical proposition.

Thus, we had to rely on natural aeration during the composting process. Composting occurs at a much slower rate with natural aeration since the amount of oxygen available to bacteria in the compost pile is restricted. Frequent turning can speed up the composting process by as much as 25% (Taiganides 1977). Turning also prevents the fermentation process from going anaerobic. The anaerobic fermentation process is slower than the aerobic process and additional problems arise with the concomitant production of hydrogen sulfide gas.

Another problem was lack of moisture control. The optimum moisture content for materials used for composting is between 50 and 60% (Poincelot 1974). Above this range, anaerobic fermentation can occur, while below this range, the composting process is slowed down dramatically. Although we felt the manure and stall litter were within or slightly above the optimal water content range, we had to rely on rainfall to provide additional moisture.

Since the compost materials were a heterogenious mixture of manure, wood shavings, urine and sometimes hay and straw, it was not possible to regulate the carbon:nitrogen (C:N) ratio of the compost. Carbon is used by bacteria for growth and energy with the portion used as energy being released as CO_2. The weight loss of compost due to CO_2 loss ranges from 47 to 80% (Taiganides 1977). While carbon is being metabolized, nitrogen is fixed within the cells of the microorganisms. Therefore as carbon is lost, the amount of nitrogen is concentrated in the compost. The optimum initial C:N range is from 30 to 50 parts carbon and 1 part nitrogen. A higher ratio may result in delay of the composting process while a lower ratio may result in a nitrogen loss through ammonification. The final C:N range after composting should be between 12 to 27 parts carbon to 1 part nitrogen. If the final C:N ratio is high, bacteria will degrade the carbon remaining in the compost at the expense of the nitrogen in the soil. Nitrogen content

in the finished compost should range between 1.2 to 1.5% on a dry weight basis (Taiganides 1977) or 80-139 ppm of nitrate nitrogen on a wet basis (Rhue et al. 1980).

METHODS

When the decision was made to transform the manure dump into a composting area, a one-year accumulation of manure and stall litter was already randomly scattered about the dump area in piles and windrows. Tents and temporary stalls had just been removed from an adjacent field leaving a 2-week accumulation of manure and stall litter from approximately 1,000 horses. Since much of the old manure had already been worked by flies and had composted in place, it was decided to layer the fresh manure from the tents between two layers of the old manure.

A bulldozer was used to spread half of the old manure and stall litter into a long pad. The fresh manure and stall litter was spread over the base pad and the remaining old manure and litter was used to cap off the pile.

Recommendations were made to fog adult flies in the composting area to knock down the existing high population. Resmethrin (Penick 1382) was to be applied with a small thermal fogger once daily in the early morning hours to minimize drift. Adult house and stable flies had been monitored for five months prior to composting using the sticky traps of Williams (1973). This monitoring system was continued after the composting process began. Immature fly populations were monitored by collection of pupae on and around the compost pile. As a check a farm, located 10 km from the test facility, housed ca. 20 horses and was chosen because of its consistently high stable fly populations. Flies were also monitored with sticky traps.

RESULTS

The finished compost pile was a wedge 15 m wide, 76 m long and 5 m high. The wedge shape was chosen so trucks bringing manure and stall litter could back up the sloping surface and pile their contents near the top of the slope. Piles were spread down the slope daily with a front-end loader. Although the weight of the machinery on the compost pile tended to compact the compost, the light texture of the mixture prevented complete compaction, and the fermentation process remained aerobic.

The initial cost of building the pile was the salary paid to a bulldozer operator for 3 days and the cost of using the machine during this time. The dump trucks, front-end loaders and their operators were involved in manure cleanup in the temporary stall area and would have been moving manure to the compost area whether or not the compost pile was being built. The daily maintenance charge is the cost of 1 man and a front-end

loader for the time necessary to spread the newly-dumped piles of manure and stall litter every day. A system for bringing manure and stall litter to the compost site had been in operation on a regular basis and composting did not increase the operating costs of this system.

Temperatures from 10 to 60 cm below the surface of the pile averaged 48°C with a range of 41 to 62°C. This is indicative that moisture and oxygen contents were within the optimum ranges necessary to support the growth of thermophilic bacteria (Poincelot 1974).

Another benefit of the high temperatures was the reduction of fly breeding at subsurface levels. As the temperature of a breeding medium approaches 50°C, fly larvae begin to migrate out; at 54°C, larvae die within 1 min. and at 60°C, death is instantaneous (Allnut 1926). Thus, fly breeding was restricted to the surface of the pile which increased the chances of larval and pupal mortality from predation, parasitization and climatic conditions. Numerous species of spiders, predaceous mites, ants, earwigs, predaceous beetles, dragonflies and predaceous and parasitic wasps, as well as many species of birds, lizards and small mammals were noted in and around the compost pile.

Adult populations of horse and stable flies decreased, resurged slightly and then further decreased over the next 9 weeks (table 1). Populations of eye gnats, which were too numerous to count prior to composting, were reduced to inconspicuous levels.

Table 1.--Weekly sticky trap catches of adult stable and house flies at the compost area before and after composting was initiated

Weeks	Stable flies		House flies	
	Compost area	Check farm[1]	Compost area	Check farm[1]
		Prior to composting		
1	1256	4840	288	40
2	1384	3948	112	20
		After composting		
3	640	3880	32	84
4	1176	3948	272	52
5	1080	3363	488	49
6	964	4516	472	40
7	728	3600	392	24
8	76	1600	410	20
9	696	2036	248	76
10	264	2032	40	172

Table 1.--Weekly sticky trap catches of adult stable and house flies at the compost area before and after composting was initiated--Continued

Weeks	Stable flies		House flies	
	Compost area	Check farm[1]	Compost area	Check farm[1]
11	87	3868	118	604
12	1448	7536	328	168

[1]The Check Farm, located 10 km from the test facility, housed ca. 20 horses and was chosen because of its high stable fly populations.

DISCUSSION

Immediately after building the compost pile, there was a drop in fly populations at the dump due to the total disruption of the habitat (table 1). This was only temporary, however, and populations began to resurge by the following week. Flies continued to breed in material around the edge of the compost pile and, until it was thoroughly worked, populations continued to remain relatively high.

The most evident indicator of fly control at the compost site was the sticky trap counts. However, trap counts can be misleading if nothing is known of the origin of the trapped flies. Migration studies at the test facility showed that the major direction of stable fly movement was from the eastern perimeter of the facility towards the compost site located on the western side of the facility. This distance, ca. 3.2 km, was covered in less than 8 hrs. Stable flies also migrated east from the compost site, but this migration was considered secondary to the one just described. Therefore, adult stable fly populations at the compost site were a combination of flies that eclosed at the site and flies that migrated to the site. This was also the case at the permanent stables 1.6 km east of the compost site. Migration studies indicated that stable flies were migrating to the stables from both sides of the test facility, with larger numbers arriving from the east. After the initiation of composting procedures and stable fly populations at the compost site decreased during weeks 3-8, populations at the permanent stables remained relatively high because of continued large numbers of flies migrating from the east (table 2).

House flies that eclosed at the compost site tended to remain at the site. There was no evidence found to indicate that house flies migrated across the test facility.

As another indication of the mobility of adult fly populations, house and stable flies were found breeding at the check farm only once during the test period. Manure and stall litter from approximately 20 horses was either stacked in compost piles or spread in the practice ring. Predators, particularly ants, prevented flies from breeding in the compost piles. It was therefore assumed that nearly all of the flies at the check farm were migrating from surrounding horse farms. Stable fly counts on horses at the check farm were frequently as high as 25 flies per leg. The lack of breeding material at the check farm probably accounted for the low house fly population (table 1).

Table 2.--Weekly sticky trap catches of adult stable flies from three different locations on the test facility before and after composting was initiated

Weeks	Compost site	Permanent stables	Eastern perimeter
		Prior to composting	
1	1256	1506	1754
2	1384	1141	1504
		After composting	
3	640	1120	1936
4	1176	1474	2780
5	1080	1859	3886
6	964	2180	4045
7	728	2712	4680
8	76	1294	2000
9	696	2650	3004
10	264	1850	3548
11	87	667	901
12	1448	3450	7752

Few stable fly larvae were found at the compost site after composting began. Most larvae seen were house flies which were active in and around accumulations of manure on the surface of the compost pile. Aggregations of stable or house fly pupae have yet to be found on the compost pile although extensive searches have been conducted on numerous occasions. The few solitary pupae that have been found were all less than 15 mm beneath the surface of the compost pile.

Evidence that stable and house flies were eclosing from the compost pile was corroborated by the presence of the immature stages of a red-orange erythraeid mite which were attached to flies captured on the sticky traps. The species of mite is unknown, but it is believed to be parasitic in the immature stages and possibly predaceous as an adult. Adults have been

found in manure piles throughout the peninsular section of Florida. Immatures are thought to attach themselves to flies soon after the flies eclose from their pupal cases. These mites are a natural marker and are an indication that flies have bred in a permanent or semi-permanent habitat of decomposing organic material. Of the flies trapped at the compost pile, house flies carrying the erythraeid mites outnumbered stable flies carrying the same species of mite by a ratio of 10:1.

The hot, dry weather which persisted during and after the initiation of composting was a contributing factor in the gradual reduction of stable and house fly populations. Occasional rains moistened the compost pile but the continual addition of fresh material to the top of the pile inhibited fly breeding.

There was a resurgence of fly populations at the compost site by week 12. This was caused by a temporary interruption in maintenance of the pile which was reflected by the resurgence of stable and house flies during week 12 (table 1). Although fogging was suggested, it was never accomplished due to a shortage of personnel. This may have been an unexpected windfall, however, since personnel at the test facility have not come to be dependent upon pesticides as a substitute for poor management.

QUALITY OF THE COMPOST

The quality of the compost is decreased by the myriad of items thrown into the pile which will not compost such as old horse shoes, 2nd and 3rd place show ribbons, beer cans and so on. These items present a problem when the compost is used since they must be separated from the compost and properly disposed of. This problem could be minimized by screening the compost before use.

Although the compost has been used with good results as a thin mulch over newly-seeded bahia grass, extensive use of the compost has been limited due to the high salt contents resulting from the high salt intake of the horses (table 3). Nitrogen is in the optimum range and phosphorous is slightly high as is the pH. These high salt values may decrease as the compost ages and the porous nature of South Florida soils may allow the salts to leach through before they become a problem to plants. Further investigation into this problem is underway.

Table 3.--Results when 1-year-old compost from the test facility was subjected to Greenhouse and Potting Media Analysis

Analysis	Optimum range[1]	Compost
NO_3-N	80-139	95.00
K	110-179	1589.00
P	8-13	58.30
Mg	60-99	59.60
Ca	140-219	80.00
Sol. salts	1000-1500	3665.00
pH	5.8-6.8	7.51 (N=9)

Note: All values expressed as ppm.
[1]As determined by the Florida Cooperative Extension Service, IFAS, University of Florida, Gainesville, FL 32611.

REFERENCES

Allnut, E. B.
1926. Some experiences in the control of fly breeding. J. Royal Army Med. Corps 47: 105-120.

Morrison, F. B.
1966. Feeds and feeding. 696 pp. The Morrision Publishing Co., Clinton.

Poincelot, R. P.
1974. A scientific examination of the principles and practice of composting. Compost Sci. 15(3): 24-31.

Rhue, R. D.; Kidder, G.; Ingram, D. L.; and Midcup, J. T.
1980. The greenhouse and potting media test. Notes in Soil Science. Univ. of Fla. Coop. Ext. Serv., IFAS, Gainesville, Fla.

Taiganides, E. P.
1977. Composting of feedlot wastes. In E. P. Taiganides (ed.), Animal Wastes, pp. 241-251. Applied Science Publishers, London.

Williams, D. F.
1973. Sticky traps for sampling populations of Stomoxys calcitrans. J. Econ. Entomol. 66: 1279-1280.

EVALUATION OF STICKY PYRAMID TRAPS FOR CONTROL OF FACE FLIES

R. W. Miller and L. G. Pickens

U.S. Department of Agriculture

INTRODUCTION

The Livestock Insects Laboratory, Beltsville, Maryland has investigated the use of sticky pyramid traps to control face flies for the past two fly seasons. These traps were evaluated on an area-wide basis as part of a face fly control pilot test study. It was our initial belief that sticky pyramid traps would not be a practical method of controlling face flies because of the labor and expense required to service them. They were, however, used in this study to test whether or not enough flies could be drawn to and captured on them to reduce field populations.

MATERIALS AND METHODS

The traps we tested were constructed as four-sided pyramids with a slope of 60° and a side height and a base length of 60 cm. The pyramids were set on top of open-ended boxes 44 cm wide and 61 cm high. Both boxes and pyramids were painted with glossy white latex paint (No. 7200, Colony Paints). The pyramids were covered with sheets of clear, colorless, 0.005 cm thick plastic to which Tack-Trap adhesive was applied.

For the portion of the pilot test reported here, concerned with the testing of the traps, farms in Howard Co., Maryland were surveyed for face fly populations in the summer of 1978 by making face fly counts on survey pyramid traps and the faces of cattle. Approximately 3 survey traps were put on each farm into pastures containing cattle. The traps were counted twice a week from June 1 until October 30.

In 1979 a group of 4 farms was designated as an untreated check area and an area containing 8 other farms was designated as the trapped area. During March and April, ca. 20, 61 x 61 cm white painted plywood panel style traps on metal stakes were placed on each of the farms in the trapped area. These

traps were covered with plastic and Tack-Trap and changed twice a week. Preliminary results suggested that two panels would catch approximately the same number of face flies as one pyramid trap. It soon, however, became obvious that this was not true under conditions of this test. We, therefore, converted all panel traps to pyramid traps by June 1. The total number of pyramid traps in the trapped area averaged 9.2 per farm or one for every 5 head of pastured cattle. The traps were serviced twice a week until the end of October.

In 1980 we put out pyramid traps on the same group of farms, but expanded the test area to include a total of 17 farms. On these farms we had an average of 13 traps per farm or 1 trap for every 3 head of cattle. The traps were in place by March 15 and were maintained until the end of October.

In 1979 and 1980, face flies on survey traps were counted as in 1978. Counts of face flies were expressed as the average number caught per day and as an index of fly activity calculated by dividing weekly 1979 and 1980 counts by weekly 1978 counts. The index was used to adjust for inherent differences in fly populations between the two areas. Face flies were also counted on the faces of 15 cattle from herds on the original farms once a week. In addition, face fly numbers on cattle were rated by the number of weeks that they attained an average of more than 10 flies per face.

RESULTS

Face Fly Counts on Survey Traps and Faces of Cows

1978	Trapped Area	Check Area	Test of[a/] Sign.
Avg. survey trap catch/day	22.7	33.7	P<.01
Avg. no. face flies/face	5.6	5.0	n.s.
No. weeks > avg. 10 flies/face	2	1	-
1979			
Avg. survey trap catch/day	47.5	94.5	P<.01
Ratio 1979 to 1978 trap catches	4.0	5.1	n.s.
Avg. no. face flies/face	5.1	4.8	n.s.
No. weeks > avg. 10 flies/face	4	4	-
1980			
Avg. survey trap catch/day	44.6	111.9	P<.01
Ratio 1980 to 1978 trap catches	3.4	5.0	P<.05
Avg. no. face flies/face	5.6	6.6	n.s.
No. weeks > avg. 10 flies/face	2	7	-

a/ Data analyzed by analysis of variance, n.s. = (P>.05).

DISCUSSION

If it can be assumed that the pyramid survey traps are a more sensitive method of measuring face fly populations than are face counts on animals (Pickens et al. 1977), the trapped area had a lower population of face flies in all three years than the check area. However, the difference in the index values was only significant for 1980. There was also a trend for lower face counts in the trapped area in 1980 as face flies only exceeded 10 flies per face two weeks during the season. This reduction in face fly populations, however, required an average of one trap for 3 head of cattle. Whether or not one would want to consider using this method of control could be dictated by economic considerations. We estimate for the plastic and Tack-Trap to treat the pyramids in the trapped area we spent $4,200 in 1979 and $12,000 in 1980. On a per cow basis, this would amount to $10 and $20 in 1979 and 1980, respectively.

REFERENCE

Pickens, L. G.; Miller, R. W.; and Grasela, J. J. 1977. Sticky panels as traps for *Musca autumnalis*. J. Econ. Entomol. 70: 549-552.

LABORATORY TESTS OF RAVINIA LHERMINIERI AS A PREDATOR OF THE FACE FLY

Lawrence G. Pickens

U.S. Department of Agriculture

When larvae of Ravinia lherminieri (Robineau-Desvoidy) were placed in cups of bovine feces which contained face fly larvae, musca autumnalis De Geer, few or no face fly pupae were produced.

Since R. lherminieri had not previously been reported as being a predator of face fly larvae, I conducted tests to determine their ability and prediliction to act as a face fly predator ("Laboratory Tests of Ravinia lherminieri (Diptera:Sarcophagidae) as a Predator of the Face Fly," Can. Entomol., in press). At 27°C, each R. lherminieri female mated 2 days after adult eclosion and produced an average of 14 larvae every 7 days over 22 days for a total of 46 larvae. An average of 63% of the larvae produced adults when they were fed only feces, compared with 89% when face fly larvae were present in the feces. The average period from adult to adult was 23 days and the average female lifespan was 51 days.

R. lherminieri larvae were most effective as predators when they were larviposited onto feces at the same time as face fly eggs were deposited on the feces.

R. lherminieri larvae significantly reduced ($P<0.05$) the production of face fly pupae at R. lherminieri-to-face fly egg ratios of 10:400 in 400 ml of bovine feces. However, they were ineffective in 800 ml of feces even at a ratio of 10:100, so the area of search appears to be very limited.

Since R. lherminieri is cosmopolitan in distribution, but is seldom numerous, it may be heavily preyed upon by other coprophilous insects, but almost nothing is known of its biology in the field. It would, therefore, appear to merit further investigation as a face fly biocontrol agent.

INSECT PARASITES OF THE HORN FLY AND FACE FLY IN MISSOURI

Gustave D. Thomas

U.S. Department of Agriculture

The purpose of these studies was to determine the native species of insect parasites of horn flies and face flies in Missouri and their efficacy as natural control agents.

HORN FLY PARASITISM STUDY

Review of Literature

The 1st report of parasitic insects attacking the horn fly, Haematobia irritans (L.) (Diptera: Muscidae), in the United States was that of Ashmead (1894). He stated that C. V. Riley had reared Spalangia haematobiae Ashmead (Hymenoptera: Pteromalidae) from this fly. Lindquist (1936), in Texas, reared 2 species of parasites, S. endius Walker (as S. muscidarum stomoxysiae Girault) and S. drosophilae Ashmead, and opined that S. endius might be valuable in the biological control of H. irritans. Bruce (1964) listed 9 hymenopterans that had been reported from the horn fly. Depner (1968), in Canada, reared 8 species of parasites, and Combs and Hoelscher (1969) recovered 7 species while working on overwintering horn flies in Mississippi.

Methods and Materials

The horn fly study was conducted for 3 years using 7 beef heards (Thomas and Morgan 1972). The pastures were visited weekly from April to November and 10 to 20 pats of fresh manure were marked with cloth flags at each visit. Flagged pats were left in the field for a week; then, they were dug up and brought to the laboratory. At the laboratory, agitator-type washing machines were used to recover the horn fly pupae from the cow dung. The collected horn fly pupae were held in glass vials in the laboratory until all the flies and parasites emerged.

Results and Discussion

During the 3 years of this study, 11 species of insect parasites were reared from the horn fly (Thomas and Morgan 1972): 10 species of Hymenoptera and 1 of Coleoptera (table 1). The braconid is a gragarious, larval-pupal parasite; female adult parasites lay their eggs in horn fly larvae and adult parasites emerge from the horn fly puparia and more than 1 parasite develops on a single host pupa. The cynipid and 2 figitids are solitary larval-pupal parasites; only a single parasite develops on a single host pupa. The diapriid and 5 pteromalids are all solitary, pupal parasites; the female adult parasites lay their eggs in the horn fly pupae and the adult parasites emerge from the horn fly puparia. The staphylinid is a solitary, pupal parasite which undergoes a hypermetamorphic development; the eggs, deposited in or around the dung pat, hatch into active 1st-instars which search out host puparia, gnaw a hole in them, enter, and feed on the host pupae, meanwhile transforming into a grublike stage.

Table 1.--Insect parasites of the horn fly in Missouri

Braconidae
Aphaereta pallipes (Say)
Cynipidae
Pseudeucolia sp.
Figitidae
Figites sp.
Neralsia hyalinipennis (Ashmead)
Diapriidae
Trichopria haematobiae? (Ashmead)
Pteromalidae
Eupteromalus sp.
Muscidifurax raptor Girault and Sanders
Spalangia haematobiae Ashmead
Spalangia nigta Latreille
Spalangia nigroaenea Curtis
Staphylinidae
Aleochara bipustulata? (L.)

The most common parasite of the horn fly in Missouri is *Spalangia nigra* (table 2). This species occurred in 22% of the pupal samples obtained in this study. A pupal sample, as used here, means the number of horn fly pupae recovered from the total number of pats marked at any 1 pasture at any 1 time. For example, in the first year of this study, we marked 20 pats of fresh dung at each farm each week. So, a pupal sample was the total number of pupae collected from 20 pats. *Spalangia nigroaenea* was 2nd and *S. haematobiae* 3rd. The other 8 species of parasites of the horn fly were recovered quite infrequently (occurred in less than 3% of the pupal samples).

Table 2.--Three most common species of horn fly parasites in Missouri

Species of parasite	Frequency of occurrence (%)
1. *Spalangia nigra*	22
2. *Spalangia nigroaenea*	10
3. *Spalangia haematobiae*	7

Percentage parasitism by all species of parasites of any one pupal sample was usually low (table 3); there was a total of 499 pupal samples in this study. In 89% of the pupal samples, parasitism by all species was 10% or less. Furthermore, in 96% of the samples, the percentage parasitized was 25% or less.

Table 3.--Frequency of 2 levels of parasitism by all species of parasites of the horn fly in Missouri

Level of parasitism	Frequency of level (%)
10% or less	89
25% or less	96

During the 3 years of this study, seasonal parasitism by all species for all farms was low (table 4). Of the 30,634 horn fly pupae collected, only 4.6% were parasitized.

Table 4.--Seasonal parasitism of the horn fly in Missouri by all species of parasites

Year	Number of horn fly pupae recovered	Percentage parasitized
1	8920	3.7
2	15005	4.1
3	6709	6.6
	30634	4.6

Conclusions

Parasitism of horn flies in Missouri is not high enough to substantially reduce horn fly populations. It appears that the parasites attacking the horn fly in Missouri are primarily parasites of other species of insects and are only incidental on the horn fly. Furthermore, there does not seem to be a specific horn fly parasite in the State.

FACE FLY PARASITISM STUDY

Review of Literature

The 1st report on the parasites of the face fly, Musca autumnalis DeGreer (Diptera: Muscidae), in the United States was that of Blickle (1961). He found that 16.3% of 2111 puparia were parasitized by Aphaereta pallipes (Say) (Hymenoptera: Braconidae) Xyalophora quinquelineata (Say) (Hymenoptera: Figitidae), and Eucoila sp. (Hymenoptera: Cynipidae). Benson and Wingo (1963) were the first to investigate the parasites of the face fly in Missouri. They reported parasitism by A. pallipes ranged from 24-84% in face fly larvae collected in late August and September. Houser and Wingo (1967) investigated the biology of A. pallipes in Missouri. They reported that seasonal parasitism of the face fly varied from 0.2-11.9%, and that the heaviest parasitism was obtained in September. Sanders and Dobson (1966) reared only A. pallipes from the face fly in Indiana.

Methods and Materials

The face fly study was conducted for 2 years using 8 beef herds (Thomas and Wingo 1968). The pastures were visited weekly from April to September and face fly eggs from a laboratory colony were used to artificially infest fresh cow manure at each visit. The infested manure was contained in pie pans which were located in large enamel pans containing soil and the seeded manure was protected from disruption by rain by clear plastic shelters. Each week the face fly pupae were collected from the soil in the enamel pans by sifting and brought to the laboratory and held in petri dishes until all the flies and parasites emerged.

Results and Discussion

During the 2 years of this study, 5 species of insect parasites were reared from the face fly (Thomas and Wingo 1968): 4 species of Hymenoptera and 1 of Coleoptera (table 5). The 2 species of braconids are both larval-pupal parasites; Alysia ribibunda Say is a solitary parasite and Aphaereta pallipes, which also attacks the horn fly, is gregarious. The cynipid is also a solitary, larval-pupal parasite. The pteromalid, Muscidifurax raptor, which also attacks the horn fly, and the

staphylinid Aleochara bimaculata (Gravenhorst), are solitary, pupal parasites. Like the horn fly parasite, Aleochara bipustulata?, A. bimaculata also undergoes a hypermetamorphic development.

Table 5.--Insect parasites of the face fly in Missouri

Braconidae
Alysia ribibunda Say
Aphaereta pallipes (Say)
Cynipidae
Eucoila impatiens (Say)
Pteromalidae
Muscidifurax raptor Girault and Sanders
Staphylinidae
Aleochara bimaculata (Gravenhorst)

The most common parasite of the face fly in Missouri is Aphaereta pallipes (table 6). This species occurred in 30% of the pupal samples obtained in this study. Eucoila impatiens was 2nd and Aleochara bimaculata 3rd. The other 2 speceis of parasites were each recovered from only a single pupal sample.

Table 6.--Three most common species of face fly parasites in Missouri

Species of parasite	Frequency of occurrence (%)
1. Aphaereta pallipes	30
2. Eucoila impatiens	26
3. Aleochara bimaculata	10

Percentage parasitism by all 5 species of parasites of any one pupal sample was usually low (table 7); there was a total of 182 pupal samples in this study. In 81% of the pupal samples, parasitism by all species was 10% or less. Furthermore, in 90% of the samples, the percentage parasitized was 25% or less.

Table 7.--Frequency of 2 levels of parasitism by all species of parasites of the face fly in Missouri

Level of parasitism	Frequency of level (%)
10% or less	81
25% or less	90

During the 2 years of this study, seasonal parasitism by all species for all farms was low (table 8). Of the 75,896 face fly pupae collected, only 3.8% were parasitized.

Table 8.--Seasonal parasitism of the face fly in Missouri by all species of parasites

Year	Number of face fly pupae recovered	Percentage parasitized
1	17412	7.8
2	58484	2.6
	75896	3.8

Conclusion

Parasitism of face flies in Missouri is not high enough to substantially reduce face fly populations.

REFERENCES

Ashmead, W. H.
1894. A synopsis of the Spalangiinae of North America. Proc. Entomol. Soc. Wash. 3: 27-37.

Benson, O. L., and Wingo, C. W.
1963. Investigations of the face fly in Missouri. J. Econ. Entomol. 56: 251-258.

Blickle, R. L.
1961. Parasites of the face fly, Musca autumnalis, in New Hampshire. J. Econ. Entomol. 54: 802.

Bruce, W. H.
1964. The history and biology of the horn fly, Haematobia irritans (Linnaeus); with comments on control. N. C. Agric. Exp. Stn. Tech. Bull. 157: 33 pp.

Combs, R. L., and Hoelscher, C. E.
1969. Hymenopterous pupal parasitoids found associated with the horn fly in northeast Mississippi. J. Econ. Entomol. 62: 1234-1235.

Depner, K. R.
1968. Hymenopterous parasites of the horn fly, Haematobia irritans (Diptera: Muscidae), in Alberta. Can. Entomol. 100: 1057-1060.

Houser, E. C., and Wingo, C. W.
1967. Aphaereta pallipes as a parasite of the face fly in Missouri, with notes on laboratory culture and biology. J. Econ. Entomol. 60: 731-733.

Lindquist, A. W.
1936. Parasites of the horn fly. J. Econ. Entomol. 29: 1154-1158.

Sanders, D. P., and Dobson, R. C.
1966. The insect complex associated with bovine manure in Indiana. Ann. Entomol. Soc. Am. 59: 955-959.

Thomas, G. D., and Morgan, C. E.
1972. Parasites of the horn fly in Missouri. J. Econ. Entomol. 65: 169-174.

Thomas, G. D., and Wingo, C. W.
1968. Parasites of the face fly and two other species of dung-inhabiting flies in Missouri. J. Econ. Entomol. 61: 147-152.

THE INFLUENCE OF PARASITES, PREDATORS, AND COMPETITORS ON HORN FLY POPULATIONS

R. L. Harris

U.S. Department of Agriculture

The horn fly, Haematobia irritans (L.), is subject to attack by many parasites and predators during its developmental stages in cattle droppings. In addition, other insects, such as scarabs, compete for the dung. Thomas and Morgan (1972) and Blume et al. (1970) have demonstrated that biological agents reduce the number of horn flies emerging from cattle dung by 80 to 95% in Missouri and Texas and that predators are the most important natural enemy of the flies. The contributions of each class of predators has not been identified; however, staphylinid beetles are the most numerous predator in the United States. We have shown that Philonthus flavolimbatus Erichson is an excellent predator of horn fly eggs (Harris and Oliver 1979). The data presented by Thomas and Morgan (1972) also indicate that staphylinid beetles are responsible for much of the mortality of immature horn flies. Although mites, histerid beetles, and predatory Diptera are also commonly associated with cattle droppings, their impact on the horn fly population has not yet been evaluated.

We surveyed the natural enemies of the horn fly on a farm near Cooks Point, Texas for the past 3 years and consistently found that the numbers of horn fly pupae recovered correlated negatively with numbers of staphylinids present. Other natural enemies did not show a consistent correlation with the number of horn fly pupae. The high negative correlation of staphylinid beetles with horn fly pupae may indicate that the staphylinid beetles reduce the horn fly population by predation. The numbers of scarabs correlated positively with the numbers of horn flies, indicating that scarabs probably have little influence on horn fly populations. The positive correlation of scarabs and horn flies is probably due to the effect of climatic and edaphic variations on both populations.

The rate of parasitism of horn flies by pupal parasites was usually less than 20%; however, the work of Combs and Hoelscher

(1969) in Mississippi and our work in Texas as well as Lindquist's earlier (1936) work showed rates of parasitism as high as 40%. The overall rate in our tests was 2.7% in 1979 and 8.1% in 1980, with an average of 4.4%. The rate was highest (43%) during the week of July 11, 1980. There appeared to be no seasonal variation, as parasitism occurred more or less uniformly from April through October. The dominate species of pupal parasites were Spalangia nigroaenea Curtis (68% of the total) and Spalangia cameroni Perkins (23% of the total). Other parasites found were Spalangia nigra Latreille, Spalangia haematobiae Ashmead, Trybliographia sp., Figites sp., and Neralsia sp. Depner (1968) and Peck (1974) found similar parasites and rates of parasitism of the horn fly in Canada. Although the overall rate of parasitism is usually low, it may have some influence on fly populations. The development of methods by which papal parasites can be made more effective is an attractive area for research. Experiments to determine if inundative releases of parasites aid in the control of the horn fly have not been conducted, and until data on the parasite effectiveness is available, this method of control remains questionable.

The importation of biological agents for the control of the horn fly in recent years has been limited to dung-burying scarabs. Three species have been released in Texas. Onthophagus gazella F., Euoniticellus intermedius (Reiche), and Onthophagus bonasus (F.). Onthopagus gazella was released several years ago and is now well established in the southern-half of Texas (Blume and Aga 1978). During peak activity, these scarabs remove over 80% of the dung from the soil surface. Euoniticellus intermedius was released near Fredericksburg, Gllespie Co., Texas, in 1977 and are presently well established (R. R. Blume, personal communication). Euoniticellus intermedius and O. bonasus were released at a farm near Cooks Point, Texas during the summer of 1980 (G. T. Fincher, personal communication) and are expected to establish themselves, for the climate at this site is milder than the climate at Fredericksburg. Onthophagus bonasus, from Pakistan, is the most recent introduction. Several species of dung beetles as well as certain species of parasites and predators have been released in Hawaii for the control of horn flies (Legner 1978). Many of these have become established; however, their effect on horn fly populations has not been evaluated. Horn fly populations in Hawaii are as high or higher than those occurring in Texas, indicating that biological control is still inadequate.

We need further data on the total effects of various biological control agents before we can develop an effective complex that will result in fly control, dung removal, and reduction of gastrointestinal parasites. Our research is now primarily directed toward the identification of such an ideal complex.

REFERENCES

Blume, R. R., and Aga, A.
1978. Onthophagus gazella F.: Progress of experimental release in south Texas. Folia Entomol. Mexicana 39-40: 190-191.

Blume, R. R.; Kunz, S. E.; Hogan, B. F; and Matter, J. J.
1970. Biological and ecological investigations of horn flies in central Texas: Influence of other insects in cattle manure. J. Econ. Entomol. 63: 1121-1123.

Combs, R. L., and Hoelscher, C. E.
1969. Hymenopterous pupal parasitoids found associated with the horn fly in northeast Mississippi. J. Econ. Entomol. 62: 1234-1235.

Depner, K. R.
1968. Hymenopterous parasites of the horn fly Haematobia irritans (Diptera: Muscidae) in Alberta. Can. Entomol. 100: 1057-1060.

Harris, R. L., and Oliver, L. M.
1979. Predation of Philonthus flavolimbatus on the horn fly. Environ. Entomol. 8: 259-260.

Legner, E. F.
1978. Muscidae. In C. P. Clausen, ed. Introduced parasites and predators of arthropod pests and weeds: A world review. USDA Agric. Handbook 480: 346-349.

Lindquist, A. W.
1936. Parasites of the horn fly and other flies breeding in dung. J. Econ. Entomol. 29: 1154-1158.

Peck, O.
1974. Chalcidoid (Hymenoptera) parasites of the horn fly, Haematobia irritans (Diptera: Muscidae), in Alberta and elsewhere in Canada. Can. Entomol. 106: 473-477.

Thomas, G. D., and Morgan, C. E.
1972. Field-mortality studies of the immature stages of the horn fly in Missouri. Environ. Entomol. 1: 453-459.

FOREIGN EXPLORATION, COLLECTION AND SHIPMENT OF PARASITES AND PREDATORS

H. Hoyer

U.S. Department of Agriculture

The European Parasite Laboratory (Science and Education Administration) was initially set up in 1919 in Southern France to survey and collect natural enemies of the European corn borer (Ostrinia nubilalis Hübner). With more research projects added, and a greater range of target pests to study in Northern Europe, the EPL was transferred to the Paris area in 1936.

This laboratory started work on dung breeding flies in 1962 after the face fly (Musca autumnalis) De Geer became a pest of cattle in much of the northeastern United States.

Fly population counts and comparisons in Europe were provided by J. U. McGuire and R. I. Sailer in 1962, to establish whether low numbers would suggest that the natural enemy complex of the muscid was important. With growing concern about the face fly spreading throughout the U.S., the interest in the mechanisms that in nature may control populations of manure and filth breeding diptera elsewhere, led to studies and surveys of dung beetles in Egypt and Pakistan. R. R. Blume is the U.S. researcher most closely associated with these studies. In France, official research units are more interested in the ecological side of dung removal and dispersion and related questions of environment in fragile ecosystems. It is generally known that the face fly can build considerable populations on cattle in various parts of Europe, while elsewhere it is not considered an economic pest. But little is known about the competition and the presence of predaceous insects in the dung in Europe, which are different from the dung complex in North America. However, J. J. Drea, then with the European Parasite Laboratory, recovered, in 1962-64, the staphylinid, Aleochara tristis Gravenhorst. The collection in Europe, and shipment to the U.S., of several hundred specimens of this predaceous-parasitic beetle allowed the initiation of mass production in the United States. It was liberated in large numbers and become established. The

reduction of, and impact on, fly abundance in some areas were reported by Wingo et al. in 1967, but the attraction of this staphlinid to field manure pads does not seem effective.

Another quest for the control of pasture breeding flies is to combine the introduction of natural predators and coprophagous scarab beetles. This has been the approach by Australian researchers who have studied this question in South Africa and France for about 10 years. The work in France is being done at the Commonwealth Scientific and Industrial Research Organization in Montpellier under the direction of A. A. Kirk. Although Australia's major concern is to disperse unwanted accumulations of cattle dung, it is also hoped that, through dispersal of the dung, the two most important dipterous pests, the buffalo fly, Haematobia irritans exigua, and the bushfly, Musca vetutissima, will be greatly reduced. During the early stages of the importation program only tropical species and scarab habitat reducers from similar climatic areas (Montpellier and other Mediterranean climatic areas similar to parts of Australia) were introduced. More recently, due to the success of establishment of Aleochara tristis Gravenhorst in the U.S., the Australian scientists are now very interested in the staphylinid beetle component of the dung fauna.

At the same time, work by M. M. H. Wallace et al. showed that predaceous mites, especially a species of the European macrochelids, can substantially reduce bushfly breeding; and consideration is being given to introduction of an additional mite predator. But it is most likely that G. W. Krantz of Oregon State University could contribute a great deal of information and advice on his recent efforts with mites to reduce pest fly density in the United States.

The small amount of work done on the ecology and biological control of the dung and filth breeding fly complex in Europe may well account for the small number of predators and parasites recorded, especially from hornflies, Haematobia irritans DeGeer, and face flies, Musca autumnalis DeGeer.

The European Parasite Laboratory is well located to carry out foreign exploration for and importation of predators, parasites, and competitors of dung and filth breeding pests. Our permanent position in Europe not only permits us to carry out exploration for a given target pest over a long period but also enables us to find and work in climatic habitats similar to those of the release areas in the United States.

The shipping techniques to prevent the introduction of foreign animal diseases are being developed jointly among EPL, APHIS (Animal and Plant Health Inspection Service) and cooperating U.S. scientists in SEA. In 1981, the search for biological control agents against the four major fly pests in the United States--Musca domestics L. (house flies), Stomoxys calcitrans

(L.) (stable flies), Haematobia irritans DeGeer (hornflies), and Musca autumnalis DeGeer (face flies)--should encompass the following:

(1) infestation and population density studies
(2) cultural practices favoring or disfavoring reproduction of pest flies
(3) predation, parasitization, and completion as means of fly control
(4) dung degradation by dung beetles and importation of requested species.

Due to travel restrictions and budgetary considerations work in 1981 will probably be centered in Northern France. In this region, there are 3 distinct dairy producing areas, 1 distinct meat producing area, and 1 dual-purpose area that will permit us to survey and study 5-6 different breeds of cattle. In addition, the climatic conditions and methods of production vary substantially among these areas.

CONCLUSION

Because of the growing awareness of the pest flies associated with cattle and their excrement, we must set up a program of biological control. A number of successful biological control projects in the United States were based on initial studies or surveys supported at early stages by the European Parasite Laboratory. We in Sevres are, therefore, open to suggestions and ideas to contribute to this new project. I may be contacted directly at the European Parasite Laboratory, 47, rue des Fontenelles, 92310 Sevres, France.

REFERENCES

Wallace, M. M. H.; et al.
1981. Personal correspondence.

Wingo, C. W.; Thomas, G. D.; and Nelms, N. M.
1967. Laboratory evaluation of two Aleocharine parasites of the face fly. J. Econ. Entomol. 60: 1514-1517.

COMPUTER SIMULATIONS OF THE INTERACTION OF A PUPAL PARASITE DURING AUGMENTATION RELEASES FOR CONTROL OF THE HOUSE FLY

D. E. Weidhaas

U.S. Department of Agriculture

INTRODUCTION

In recent years Morgan and coworkers have demonstrated that the control of house fly populations at poultry operations in Florida is possible by augmenting the natural abundance of a pupal parasite with sustained releases of mass-reared parasites, *Spalangia endius* Walker, (Morgan et al. 1975a, 1975b, Morgan et al. 1976, Morgan and Patterson 1977, Morgan et al. 1980). Following the first field studies, Weidhaas and Haile (1977) developed and reported a system for modeling the interaction of the host and its parasite on the basis of life history parameters and presented theoretical results of computer simulations in which the interaction of the host and parasite was studied. They considered their simulations theoretical since the life history parameters were based on laboratory rather than field data. Furthermore, there were no estimates of the absolute density of house flies in the early parasite release studies making verification of the simulations impossible. In their last report of the control of house flies by parasite releases Morgan et al. (in press 1981) reported a method and results of estimating the absolute density of house flies before and during their releases. Thus, it appeared possible to verify the model and simulation techniques with actual field data.

The purpose of this paper is to review the concepts and techniques used in simulating the interaction of the parasite and the house fly and to compare the results of simulations to the actual field data. Details of actual parasite release studies and the model and simulation methods are available in the literature cited above.

METHODS AND MATERIALS

Model and Simulation Techniques

Our model and simulation approach to following the dynamics of insect populations is based on a life history or life table analysis for each insect species involved. Figure 1 lists the six life history parameters we use to control the dynamics of the populations and develop life tables. Figure 1 also includes symbols, a numerical example for each parameter and an equation relating the parameters to the generation growth rate ($R_o = 2$). The value of the daily growth rate (λ) for a generation growth rate of 2 is included, but not derived.

PARAMETERS FOR LIFE HISTORY

Survival in immatures	S_i - Avg. daily	-0.7463
Development time of immatures	I - Days	-9
Survival of adult females	S_a - Avg. daily	- .75
Preoviposition time	D - Days	-3
Cycle of oviposition	C - Days	-1
Number of ♀♀ eggs per oviposition	M -	-16.5

$$R_o = \frac{S_i^{I} \cdot M \cdot S_a^{D}}{1 - S_a^{C}}$$

$$2 = \frac{.7463^{9} \cdot 16.5 \cdot .75^{3}}{1 - .75^{1}}$$

λ = Daily rate of increase = 1.0481296

Figure 1.--Eight life history parameters used in the model and simulations with symbols, numerical examples and equation.

Figure 2 illustrates, conceptually, both the life history approach to modeling and the computer simulation techniques we use in their simplest form. For both the house fly and the parasite we are dealing with insects that occur in overlapping generations so that all stages of the insect and all age classes are or can be present at a given time. Our problem is to move a population through time, in this case daily, by updating the numbers of each stage and in each age class each day. Figure 2 illustrates such a process for the house fly. For each calendar day we have provided storage boxes for the various age classes. Three of our life history parameters provide the information required to determine the number of storage boxes. The development time of the immatures in days sets nine storage boxes; one for eggs; five for larvae and three for pupae. The preoviposition time sets three storage boxes for adult females before they lay eggs. The cycle of oviposition in days defines which age classes of females lay

eggs. In this case we have live females laying "an average number of eggs" each day after they begin ovipositing. Rather than using a large number of storage boxes to remember all age classes of adult females until the last one dies, we accumulate egg-laying females in a single box to save storage. Multiplying the number of egg-laying females by the average number of eggs per oviposition provides the eggs for that particular day. Thus, the structure of our model and simulation is set by three of the life history parameters. Note that for simplification we keep these parameters as constants although for more comlex models they can be made variable by relating them to temperature effects. Note also that one must remember whether one is dealing with only females or both males and females and the sex ratio of the insect is a necessary parameter.

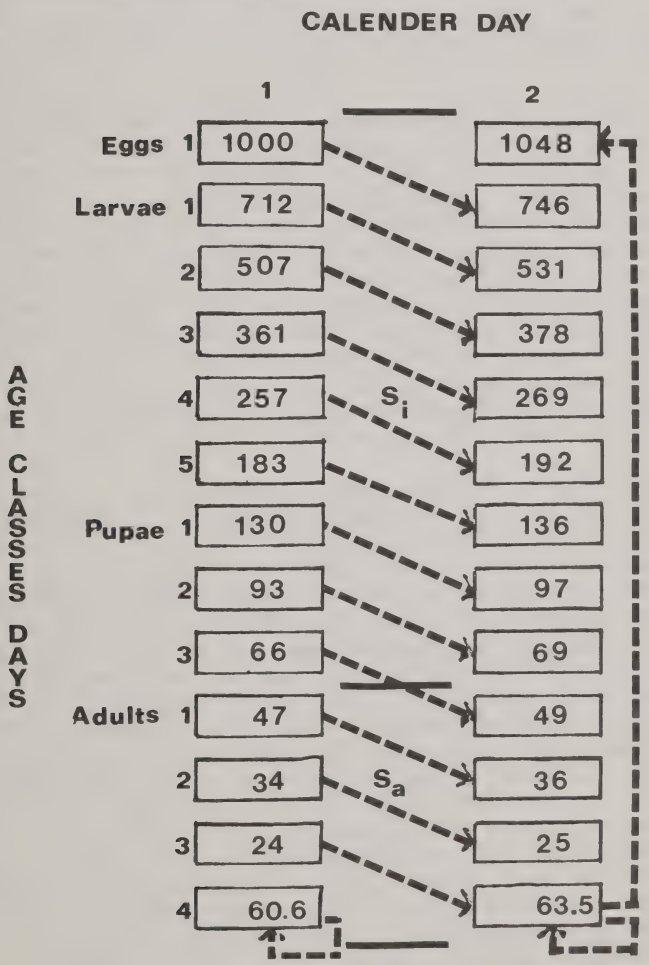


Figure 2.--Conceptualization of the model and simulation techniques in its simplest form.

With the model structure developed we have only two requirements: (1) to provide appropriate numbers for the age classes on the day a simulation is to start (initialize the model) and (2) to move the model daily through time extracting the data applicable to our purpose as it moves through time. The other three life history parameters are used to either provide or move the numbers needed for each box. If we had all of the life history data on any field population at the particular time we needed it (past, present, or future) our model could be run with no doubt of its veracity or applicability in providing solutions to problems. Unfortunately, such field data is not available or it is not applicable to the time or place it is needed. We are forced to use laboratory data for the various parameters and our best judgement on their values in the field based on knowledge and experience. This process is helped by the theoretical mathematics developed for analyzing life table data.

The initialization of our model is illustrated in Figure 2 with theoretical numbers. One chooses a starting place (in this case the number of eggs) and assigns an arbitrary number (1000) there. The numbers in all of the other boxes on that day are a function of the daily growth rate (λ) and the daily survival rate. Then, for the immatures the number in each box is obtained simply by multiplying the preceeding box by S_i/λ; for adults, by S_a/λ. The population is moved through time simply by moving one age class (or stage) to the next after adjusting it by S_i or S_a. In our system special attention is given to the mechanisms of moving 4-day-old adult females since all age groups of egg-laying females are accumulated there before multiplying by the average number of eggs per oviposition.

At this point, it is important to summarize our reasons for adopting this type of model and simulation. It utilizes the type of data that has been accumulated through biological, ecological and control studies either in the laboratory or the field. It allows analysis, synthesis and application of data within a framework that is familiar to biologists and control specialists. It provides a base for the development of more complex models and for comparison to those that use a mathematical approach. It allows the interaction of the dynamics of an insect population with the application of a variety of control methods. For example, it is easy to visualize in figure 2 that the effects of a non-residual insecticide (larvicide or adulticide) can easily be added. The only requirement would be reducing the number of larvae and/or adults by the degree of effectiveness of the treatment on the day of application. Adding additional storage space to follow the numbers of sterile and fertile males or various genotypes with programming to allow mating between the types allows simulating the effects of releases of sterile insects or genetically-altered insects. Such programming can also be visualized for attractants, pheromones, growth regulators

and biological control agents as long as the action of these agents or methods can be described. Finally, we have used this system of modeling and simulation because it allows us to control the dynamics of populations primarily by our knowledge and judgement of the actual and potential growth rates of insect populations from existing laboratory and field data. Note from figure 2 that our concept allows populations to increase, decrease or remain the same by changing only the values for survival of the immatures and adults. In most cases we use growth rates of what we consider to be reasonable magnitude to set values for S_a and S_i. Such an approach may seem to be arbitrary. However, it is generally accepted that our laboratory data defining biotic potential, i.e. maximum growth under ideal conditions, does not describe observed density changes in field populations subjected to environmental stresses. It is also true that the growth rates of field populations regulated by environmental factors would not be equal to growth rates of field populations reduced below the carrying capacity by man-imposed control strategies when density-dependent regulation comes into play. It seems logical to assume that growth rates for populations reduced below an existing carrying capacity, i.e. the net result of control or population management by factors other than naturally-existing ones, will result in population growth rates that are below the biotic potential but above those normally observed without control or management. Until the quantitation of such growth rates under a variety of environmental conditions is completed, modeling and simulation will require arbitrary decisions and remain theoretical until verified by field experimentation. Such is the nature of the process at this time and probably the strongest argument for encouraging the use of models and simulations.

Parasite--House Fly Model and Simulations

Figure 3 is a conceptualization of a model which will allow the interaction of a pupal parasite population and a house fly population. It should be considered as a description of all the stages and age classes that exist on a single day for each insect. The part describing the house fly may be different in shape, but it is the same as that described in figure 2. The part describing the parasite is constructed the same as that for the house fly except that the population parameters are different.

I = 28
D = 0
C = 1

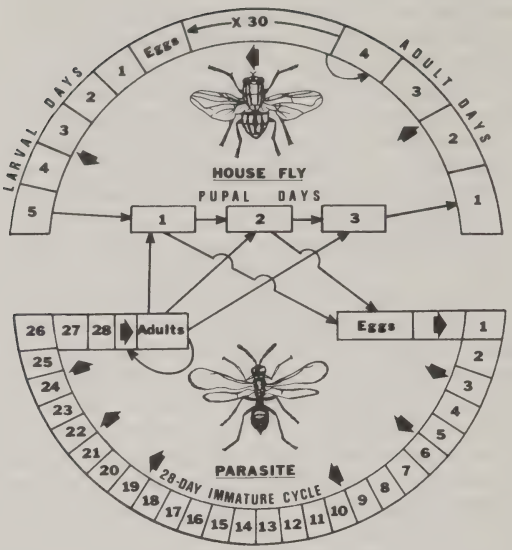


Figure 3.--A conceptual model of both the house fly and a pupal parasite.

Without repeating detail it is easy to visualize that the numbers for each age class of the two insect species can be initialized and moved through time on a daily basis as described above for the house fly. Our problem now is to provide for the daily interaction of the two species. Figure 4 highlights this interaction. Again we reiterate that we would prefer field data to describe the interactions. However, it was impossible to develop such field data and we used laboratory data for the following interactions. Female parasites must find fly pupae in which to lay an egg. The first question deals with how many pupae (on the average) one female parasite can find and parasitize in one day. In the laboratory, where searching is not difficult, this value averaged ten. Thus, multiplying the number of adult female parasites by ten provides, at least, a starting point in determining how many of the house fly pupae present in the three age classes will be parasitized on a given day. Our second question involved any preference of parasites in laying their eggs in fly pupae of different ages. Laboratory experiments showed that there was no preference. Thus, it was possible to distribute the number of pupae parasitized daily proportionally to the numbers in the three age classes of fly pupae. Our next question involved the fate of the pupae parasitized. In general, when 3-day-old pupae were parasitized an adult house fly emerged and when 1- and 2-day-old pupae were parasitized the fly was eventually killed and a parasite emerged. Thus, it

was simple to program the fate of parasitized pupae; 3-day-olds were allowed to proceed through the house fly cycle and 1- and 2-day-olds were transferred to the immature cycle of the parasite and allowed to complete development to adult parasites. The sex ratio of the parasite was shown to be two females:one male.

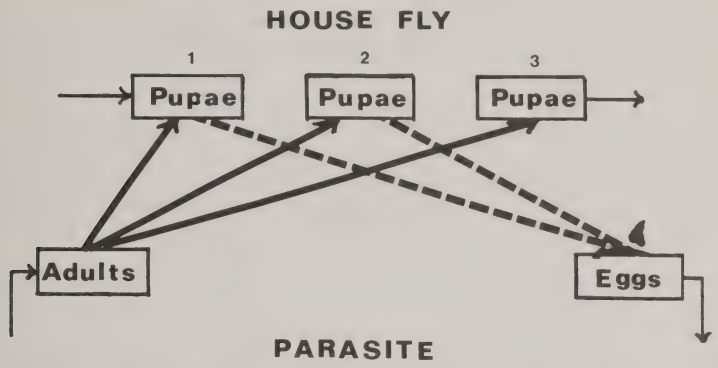


Figure 4.--Conceptualized daily interaction of a parasite population with its house fly host.

At this point our interest turns to the problem of comparing computer simulations of the interaction of a parasite and the house fly to actual field data on the control of house flies by the sustained releases of parasites. Morgan et al. (in press, 1981) published results of such a release. They made estimates of the absolute density of house flies, kept track of the number of parasites released, estimated the relative density of house flies and assayed the percent parasitism from pupae sampled in the release area. Figure 5 is a reproduction of their results showing the relative density of house flies from June 11 through July 16, 1979 when parasites were released. Figure 6 is a reproduction of the degree of parasitism they measured during the same period. For the details of this experiment the reader is referred to the original publication. In general, the density of house flies at the release site increased and decreased during the first five weeks in a pattern similar to that observed at an untreated control site several miles away. During the sixth week the population at the untreated control increased while the fly population at the release site decreased at a rate approaching the natural rate of mortality expected for the adult stage.

The decrease of adult fly density at a rate approaching natural mortality agrees with the data for percent parasitism where values varied between 90 and 100% during the last three weeks.

There were three major requirements to proceed with computer simulation of these field results: (1) absolute density

estimates of the fly population were needed to initialize the model; (2) the average number of parasites released per day was needed to input into the parasite cycle throughout the simulation and (3) values for the six life history parameters of each insect had to be selected.

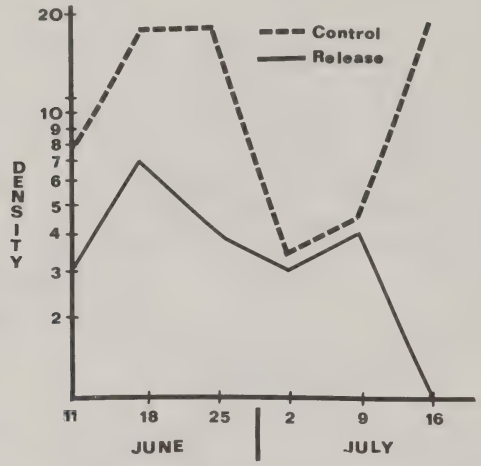


Figure 5.--Data from Morgan et al. (in press, 1981) showing the density trends of a house fly population subjected to parasite release and untreated control.

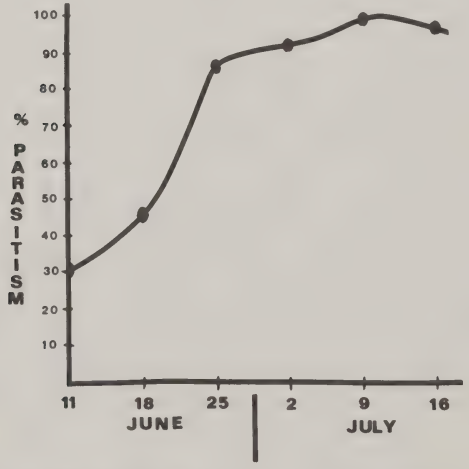


Figure 6.--Data from Morgan et al. (in press, 1981) giving the percent parasitism observed in house fly pupae during parasite releases.

Morgan and colleagues reported their estimates of the absolute number of 3rd stage larvae present at the release site when releases were started and how they made these estimates. Reference to figure 3 will help in visualizing our procedures. The total number of 3rd stage larvae were proportioned into the last three storage boxes for larvae (3rd stage larvae) as follows:

Total 3rd stage larvae = $x + x(S_i/\lambda) + x(S_i/\lambda)^2$
where x = the number of 3rd stage larvae in the first box.

The rest of the age classes were filled as described earlier. To accomplish this initialization we had to have estimates of growth rate and survival. We used a generation growth rate of two and the values in figure 1 since inspection of figure 5 indicated that the fly population was increasing at that time. Morgan reported the average number of female parasites released per day during the six-week release so that figure was available to us. The simulation was initialized with the average number of parasites released on that day and all other age classes were set at zero. To run the model daily through time we now had only to specify the survival values for each insect. For the house fly we used the values in figure 1 for a generation growth rate of two for the first week of the simulation and then reduced the values so that the generation growth rate was only one.

In summary our simulation was run by using Morgan's value of 1.53 million total 3rd stage larvae present at the start of releases, filling the remaining life history boxes for the house fly by calculations illustrated previously, releasing 125,000 female parasites each day during the simulations and controlling the dynamics of the population with the following values for the life history parameters.

	House Fly		
	Week 1	Weeks 2-6	Parasite
S_i	0.746	0.691	0.93
I	9	9	28
S_a	.75	.75	.667
D	3	3	1
C	1	1	1
M	16.5	16.5	10, 5, 1, 0.5

Figure 7 is a plot of the percent parasitism from the first two simulations in which the number of fly pupae parasitized per day per adult female parasite was set at ten in agreement with our laboratory data and then reduced to five. In both cases the results showed a much higher degree of parasitism than observed in the field. Our decision to reduce this value further and compare additional simulations seemed the logical

approach. The "effective searching ability" of the female parasite under field conditions must be much reduced over that observed in the laboratory where little or no searching is required. Figure 8 plots the percent parasitism when the effective searching ability is set at 1 and 0.5 fly pupae per day per female parasite. These results suggest that the effective searching ability lies somewhere between 0.5 and 1.0. Figure 9 repeats the field density for the house fly populations given in figure 5 and adds the density trends developed from the simulation (0.5 pupae/day/female). The agreement in population density trends appears relatively good.

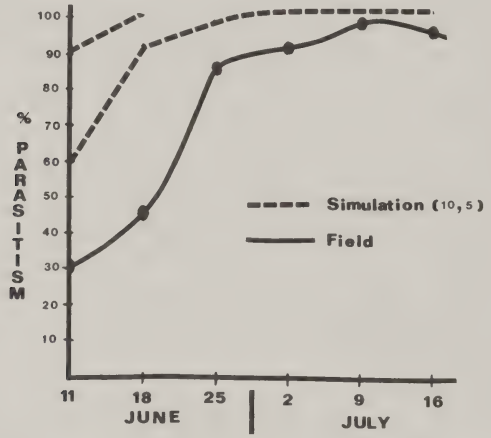


Figure 7.--Percent parasitism obtained in two simulations compared to field data of Morgan et al. (in press, 1981).

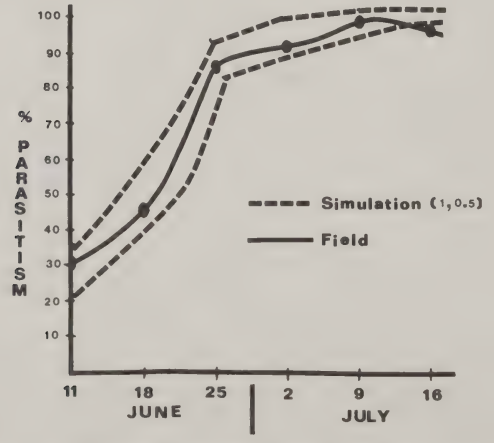


Figure 8.--Percent parasitism from two additional simulations compared to data of Morgan et al. (in press, 1981).

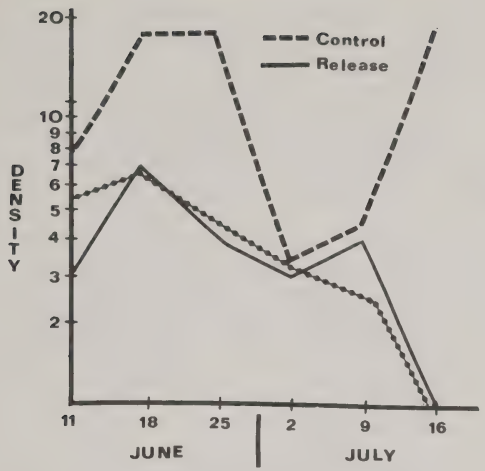


Figure 9.--Density trends of a house fly population subjected to parasite releases in computer simulations compared to field data of Morgan et al. (in press, 1981).

CONCLUSION

We have concluded that it is possible to reproduce field data by simulation techniques using the best available data for the life history parameters of the fly and parasite, their interaction and experience on the dynamics of their populations. However, it was necessary to estimate an "effective searching ability" of the female parasite under field condition by "trial and error" simulations to make the data fit. Obviously, some of the error and "difficulty in fitting" could involve less than precise estimates of other variables. For example, we have assigned an arbitrary value to the average daily survival of adult female parasites (0.667) under field conditions based on laboratory data on survival. The parasites we release are placed in the field as immatures within house fly pupae in protected containers. They are probably exposed to more hazards so that their survival is reduced either before or after adult emergence. Emergence in a non-natural habitat may not only affect survival, but it also may affect searching time or efficiency.

Until more data is available this system appears adequate for developing hypotheses and strategies for the testing and use of this parasite. For example, figure 10 uses the simulation process to project the hypothetical trends of a house fly populations (isolated) subjected to one month of parasite releases during March and the subsequent effect resulting from the F_1 and F_2 progeny from the released parasites. This example was published by Weidhaas and Haile (1977) and details are covered there. It indicates, at least theoretically,

that a full season's control of a house fly population could be obtained by just one month of parasite releases if no immigration occurred.

Month	Generation	Avg. no. ♀ flies present/day: In uncontrolled population	After 1 mo of continuous releases	% control achieved
Mar.	1	300,000	220,000	27
Apr.	2	1,500,000	300,000	80
May	3	7,500,000	500,000	95
	4	37,500,000	400,000	99
June	5	37,500,000	10,000	99.7
	6	37,500,000	50	99.99
July	7	37,500,000	500	99.99
Aug.	8	37,500,000	5,000	99.99
Sept.	9	7,500,000	50,000	99
Oct.	10	1,500,000	500,000	67
Nov.	11	300,000	300,000	0

Figure 10.--Trends of density of hypothetical house fly populations that were uncontrolled or subjected to 30 days of release of parasites at the start of the fly breeding seasons.

Hopefully, the modeling and simulation process will help to increase our understanding of the relationship between insect population dynamics and control strategies (i.e. pest control or management), identify the most important data needed to improve the process and provide working hypotheses for field testing.

REFERENCES

Morgan, P. B.
1980. Sustained releases of Spalangia endius Walker (Hymenoptera: Pteromalidae) for the control of Musca domestica L. and Stomoxys calcitrans (L.) (Diptera: Muscidae). J. Kans. Entomol. Soc. 53: 367-372.

Morgan, P. B., and Patterson, R. S.
1977. Sustained releases of Spalangia endius to parasitize field populations of three species of filth breeding flies. J. Econ. Entomol. 70: 450-452.

Morgan, P. B.; Patterson, R. S.; and LaBrecque, G. C.
1976. Controlling house flies at a dairy installation by releasing a protelean parasitoid, Spalangia endius (Hymenoptera: Pteromalidae). J. Georgia Entomol. Soc. 11: 39-43.

Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Weidhaas, D. E.; and Benton, A.
1975a. Suppression of field populations of house flies with Spalangia endius. Science 189: 388-389.
Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Weidhaas, D. E.; Benton, A.; and Whitfield, T.
1975b. Rearing and release of the house fly pupal parasite, Spalangia endius Walker. Environ. Entomol. 4: 609-611.
Morgan, P. B.; Weidhaas, D. E.; and Patterson, R. S.
1981. Host parasite relationship: augmentative releases of Spalangia endius Walker used in conjunction with population modeling to suppress field populations of Musca domestica L. J. Kans. Entomol. Soc. (In Press).
Weidhaas, D. E.; Haile, D. G.; Morgan, P. B.; and LaBrecque, G. C.
1977. A model to simulate control of house flies with a pupal parasite, Spalangia endius. Environ. Entomol. 6: 489-500.

INTEGRATED PEST MANAGEMENT--FEEDLOTS AND DAIRIES

John B. Campbell

University of Nebraska North Platte Station

The Feedlot Pest Management Project current in existence in Nebraska has been developed as a result of experience and information gained from a pilot IPM project funded by the Federal Extension Service. This pilot project was conducted in Dawson and Lincoln Counties from 1976 to 1979 in 73 feedlots and 5 dairies. The experience gained from working with this broad spectrum of animal management systems has been beneficial in conducting the current program.

We have broadened the scope of the program to include not only flies but rodent control (rats and mice) as well, and on a commodity basis to include both cattle and swine. In addition, we have incorporated two research components into the IPM program. One research component is the cost of sanitation (the main thrust of the IPM program in terms of fly control) in comparison with chemical costs and losses in animal production. The other research component is SEA/AR's evaluation of biological control (fly parasites) and the concept of insecticide-treated stable fly attractant traps.

The economics of fly and rodent control and research on new concepts of control have been integrated into one management program. County agricultural agents, wildlife management specialists, agricultural economists, agricultural engineers and entomologists from the University of Nebraska and/or SEA/AR are working cooperatively on this program. It is being conducted in 15 feedlots in Cuming County, Nebraska, a major feeding area in northeast Nebraska.

The program is funded from several sources. The major source is the IPM formula money allotted to states based on insecticide use. This money is administered by the University of Nebraska Environment Programs and is dispensed on a competitive basis. Entomology submitted a proposal for a joint crop and feedlot IPM program which was approved by the IPM Advisory Committee. This project is for a 5-year duration.

In concept, its goals is to introduce IPM into various areas of Nebraska. Nebraska IPM staff work with interested parties long enough to establish a workable IPM program then move to another location. The wildlife staff, working with rodent control, are following the same general procedure. The SEA/AR engineering and entomological input is funded by the USDA as part of their research mission. The Agricultural Economics program is funded with a grant obtained from the USDA Economics Division.

The Feedlot IPM Program is conducted from a management standpoint as follows:

1. An informational meeting, sponsored by the local feeder organization and the County Extension Service, is utilized to explain the concept and operational procedures of the IPM program. At the conclusion of this meeting, feeders interested in cooperating in the program are requested to indicate their locations on a county map.

2. Prior to the beginning of the fly season, all cooperating staff meet for training, and final details on cooperative efforts and procedures are decided at that meeting.

3. The scout(s), project leader, and scout supervisor visit each cooperating feedlot prior to the advent of the fly season. At this initial visit, the feedlots are mapped in detail. Included on the map, in addition to feedlot pens numbered according to the operator's numbering system, are any features that might influence fly breeding or fly control efforts (sick pen facilities, drainage areas, swine facilities, feed mills, silage pits, hay stacks, weedy areas, shelterbelts, and potential fly breeding areas within or near the pens). The initial visit is also used to determine, in detail, the animal waste and pest management system employed by the feedlot operator. These facts are vital when it becomes necessary to make control recommendations. As soon as possible after the initial visit, aerial photographs are taken of each feedlot. These photographs and the maps make it much easier for the scout, scout supervisor and project leader to discuss a problem and make recommendations without all three having to revisit the lot.

4. Pest population monitoring begins at a time anticipated to be slightly ahead of the fly season (usually the first week in June). Sticky traps, live animal counts and Scudder grill counts are used to monitor house and stable fly populations. Scout visits are usually at 2-week intervals (research lots are scouted weekly). In addition to pest populations, the scout records management measures such as chemical control or feedlot cleaning operations. The scout is equipped with a camera to photograph fly or rodent problem areas. These photographs can be used to consult with engineers or

other experts in determining solutions for particular problems.

5. The information gathered by the scout is placed in a computer. The computer print-out is then available to the scout supervisor, project leader, and the research unit. A copy of the print-out plus the feedlot map and recommendations are sent to the feeder. The print-out contains fly population numbers for the week of the scout visit and the past two visits in order to evaluate progress or failure in control measures. It also gives the average fly population numbers for the entire program so the operator can compare his situation with the average for the area. The computer program allows communication between the project leader and scout supervisor concerning recommendations (i.e. North Platte is 250 miles from Cuming County. It was not possible or necessary for the project leader to be in Cuming County every week. The information supplied by the scout and scout supervisor, via computer, was adequate for recommendations to be made from a remote location without actually seeing the situation.) If stable flies exceed 5 flies/leg (a number indicated by research to be economic) or if breeding areas indicate a potential for dramatic increases, control measures are suggested. These recommendations are dependent on what systems are available at a particular feedlot. Trap counts usually reflect population trends prior to fly numbers reaching the economic threshold on the animals. Recommendations for house fly control are difficult because there is little research data to indicate economic thresholds. We arbitrarily use numbers of 100/trap to recommend control because this is the level at which house flies are, at the least, a nuisance.

6. The program is terminated at the end of the fly season (usually in September). After the program is terminated, the cooperating staff and feeders are asked to evaluate the program and suggest program improvements. This information is utilized in preparing the annual IPM Progress Report required by the IPM Advisory committee and the USDA. The information is also used in planning the program for the following year.

The Feedlot IPM Program is different from crop programs for two reasons. Unlike crop programs, one season is enough for the feeder to learn to carry on his own IPM program. Many of the sanitation measures solve fly or rodent problems permanently. Most of the insecticide recommendations are such that a feeder learns to get maximum benefit from insecticide use. The feeder also learns throughout the summer to correlate animal behavior to fly population levels and subsequent economic losses. It is for these reasons that commercial agricultural consultants are not interested in the feedlot program; because if proper sanitation measures and waste

disposal systems are adopted by a feeder, consultant services are no longer needed. Further, feeder surveys indicate the feeder either feels he can learn enough to handle the situation himself or he wants control of flies included in the program. This latter requirement is basically at odds with the concept of IPM programs. A consultant involved with pest control might be reluctant to make recommendations which would eventually make control measures unnecessary. Because of these considerations, we utilize feedlot IPM as a demonstration, education process rather than an ongoing long-term program. We move into a feedlot area, establish a program and, with tours and educational meetings, demonstrate the purpose and success of the program. The following year, we move to another feeder area and repeat the process. The data accumulated and the suggestions for program improvement from the feeders and cooperating personnel enables us to prepare educational material that will make the problem of pest control at feedlots easier for feeders to solve.

INTEGRATED MULTIPEST MANAGEMENT, A PILOT PROGRAM FOR POULTRY AND LIVESTOCK IN NORTH CAROLINA

Donald A. Rutz

Department of Entomology, Cornell University

Poultry and livestock production is of great importance to North Carolina agriculture, grossing over 43 percent of the total farm income of $3.9 billion in 1979. Specifically, income from poultry ($859 million) and hog ($407 million) production in North Carolina has ranked second and third, respectively, of all farm commodities (crop and livestock) over the past several years.

A variety of pests impose serious constraints on poultry and livestock production. External parasites, filth flies, mosquitoes, biting flies, beetles, rodents, and birds all have been identified as common and significant pests of poultry and livestock, particularly in confined production facilities. Large pest populations can cause significant economic losses by having a direct adverse effect on animal health, growth, and production. In addition, some of these pests can become a severe nuisance problem both around the farm and in nearby communities. It is estimated that northern fowl mites and filth flies cause annual losses (losses and control costs) of at least $8 million to the poultry industry of North Carolina, while ectoparasites (lice and mange mites) and filth flies cause annual losses of at least $10 million to the hog industry.

Present poultry and livestock pest management techniques rely heavily on pesticides. Today, pesticides and their cost of application are a significant production expense for poultry and livestock producers. Furthermore, this single-method pest management approach, although often effective, has many serious drawbacks, including the development of pesticide resistance, destruction of natural enemies of the pests, illegal levels of pesticide residues in eggs, meat, or milk, and environmental pollution. An integrated approach to pest management in confined poultry and livestock production, using proper farm management practices, good sanitation practices, and carefully scheduled pesticide applications based on need would mitigate many of these problems.

Integrated pest management (IPM) is the selection and integration of cultural, biological, and chemical methods to manage a pest based on anticipated economic, ecological, and sociological consequences. A properly developed and applied IPM program would (1) minimize poultry and livestock losses caused by the pests, (2) optimize the use of natural enemies of the pests (parasites and predators), (3) optimize the use of pesticides, (4) minimize the development of pesticide resistance, (5) minimize pesticide residues in eggs, meat, and milk products, and (6) minimize and/or make more efficient use of producer's pest management costs.

Successful IPM programs for poultry and livestock have not evolved concurrently with the significant progress made in IPM programs for agronomic crops. To date, only five poultry and livestock IPM programs, involving stable flies and filth flies on feedlot cattle in Nebraska, pest flies on poultry in California, mosquitoes on range cattle in Louisiana, stable flies on range cattle, dairy cattle, and poultry in the Virgin Islands, and horn flies on range and dairy cattle in Hawaii, have been successfully developed and implemented (Anon. 1979).

A pilot poultry and livestock IPM extension demonstration program is currently being implemented in North Carolina. This program is directed at the management of the entire arthropod, rodent, and bird pest complex at poultry and livestock facilities.

The specific objectives of this integrated multipest management program are:

1. To develop and implement a systematic external parasite (mite, louse, bed bug), filth fly, biting fly, mosquito, beetle, rodent, and bird pest management demonstration program for use on North Carolina poultry and livestock farms.
2. To evaluate the cost effectiveness of the program.
3. To determine the practicality of establishing integrator, producer group, or private farm service organizations for continued pest management services.
4. To use the program as a training facility for individuals at all levels of operation.

AREA OF ACTIVITY

The program is being developed and tested in Duplin County in the Coastal Plain and Chatham County in the Piedmont region of North Carolina. These counties were selected for their high concentration of poultry and livestock production facilities. Caged-layer, broiler-breeder, broiler, turkey breeder, turkey fryer, swine breeder, and swine feeder producers and integrators are participating on the program.

The majority of the poultry and a large percentage of the swine in both counties are integrator owned. Integrators supply the feed, animal medicines and vaccines, and some provide insecticides and rodent baits. The producer owns the production facility and is responsible for such care of the animals as feeding, watering, and egg collecting, and is paid a contract fee for the eggs or animals produced.

METHODS AND PROCEDURES

Pest management agents, one in the Coastal Plain and one in the Piedmont, are responsible for the day-to-day operation of the program at the county level. Both agents are trained in veterinary entomology. These agents work out of the county extension office and complement the existing poultry and livestock agent.

The program operates on a year-round pest management calendar. The key element in an effective IPM program is accurate and timely pest monitoring. Pest monitoring is conducted by the pest management agent who visits each production facility either weekly or once every two weeks, depending on the severity of the pest problem and the type of facility.

Pest monitoring involves the use of various monitoring devices, an animal inspection, and a facility inspection. The agent identifies the pests, determines their stage(s) of development, assesses population levels, and, at the end of the visit, prepares a pest management report. This report contains pest status information (arthropod, rodent, and bird pest problems observed, and population numbers), facility problems (leaky waterers, feeders, etc.), and recommendations for management. The report is prepared in triplicate; one copy is left with the producer; one is sent to the integrator; and one is kept in the program file. The pest management agent also provides on-site pest management training, including training in cultural and biological pest management methods, proper insecticide dilution and application procedures, rodent baiting procedures, etc.

MANAGEMENT RECOMMENDATIONS AND MONITORING METHODS

Poultry Pests: Filth Flies (house fly, lesser house fly, black garbage fly, soldier fly, blow fly, flesh fly, stable fly, and false stable fly)

Management

Cultural Control. Manure management - Proper manure management is the key to successful filth fly management. If manure is kept dry, little or no fly breeding will occur. Several factors are involved in maintaining dry manure. These include

proper site selection for the facility, proper grading and drainage around the facility, maintenance of watering systems to prevent leaks, keeping vegetation cut around the facilities to provide good air circulation, use of fans for air circulation, and leaving an absorbent base of old manure when manure is removed from the houses. Manure should be removed from the houses only during the cold months (November through March) when the flies are inactive.

Sanitation - Remove and bury or incinerate dead birds, spoiled feed, broken eggs, or any other material that will attract and produce flies.

Biological Control. Numerous natural enemies of filth flies are present in the manure (Rutz and Axtell 1980, 1981). These natural enemies are most effective in killing flies when the manure is dry. At cleanout, do not remove all the manure. This old manure provides a source of filth fly natural enemies to reinvade the new manure.

Chemical Control. A combination of adulticides and larvicides can be used. Adulticides include baits, residual sprays on upper inside surfaces where flies rest, and space or mist sprays. Larvicides should be used ONLY for spot treatment in those areas of the manure where large numbers of fly larvae are observed. Extensive use of larvicides over the entire manure surface is NOT recommended because of harmful side effects on the natural enemies of flies active in the manure.

Monitoring

Adult Flies. Adult flies are monitored with baited traps, 3.8-liter plastic milk jugs with four 5-cm. diam. holes cut in the upper part of the sides to allow entrance of the flies, which are attracted to 25 g of Improved Golden Malrin(R) fly bait in the bottom of the jug. The traps are suspended 30 cm. from roof supports with wire. Trap number and placement varies with facility type. In high rise and wide-span, caged-layer houses, six to eight baited traps per house are used, two at equal distances at each end of the house and one or two at equal distances down each side of the house. In narrow caged-layer houses, two traps per house, one at each end of the house, are used. Three to five traps per house, placed at equal distances down the center of the house should be used in broiler-breeder facilities. The traps are left for 7 or 14 days, depending on the monitoring schedule, and then the number and species of flies collected are counted. The traps are then cleaned and fresh bait added. Although filth flies are generally not a problem at chicken broiler and turkey breeder and fryer facilities, these traps may also be used at these facilities if needed.

Treatment with adulticides is recommended when an average of 350 flies is caught per trap per week (700 flies per trap per two weeks).

Larvae. Manure is inspected for large concentrations of filth fly larvae. Spot treatment with larvicides is recommended, but ONLY on those areas where large concentrations of filth fly larvae are observed.

Poultry Pests: External Parasites (northern fowl mite, chicken mite, chicken louse, and bed bug)

Management

Cultural Control. Strict sanitation measures, elimination of hiding places by removing loose boards and rubbish, bird-proofing, and rodent-proofing are essential for effective management of poultry external parasites.

Chemical Control. Insecticides should be used ONLY when necessary. Treatment with recommended insecticides for northern fowl mites and lice should be carried out primarily on the birds but throughout the building and on the equipment for chicken mites and bed bugs.

Monitoring

For northern fowl mites and lice, the vent, legs, breast, and ventral aspect of the neck of a systematic sample (between 0.1 percent and 0.2 percent) of the birds in each house are inspected at least once every two weeks. Hot spots of mites and lice will often be observed, and are treated to prevent the spread of these external parasites throughout the flock.

Bed bugs and chicken mites can often be detected by inspecting curtain folds, nests, cracks, and crevices; however, return visits at night to inspect the birds may be necessary to verify their presence.

Treatment with insecticides is recommended when an average of 100 mites/bird or 30 lice/bird is observed. If bed bugs are present, the birds and the facility should be treated immediately.

Poultry Pests: Mosquitoes

Management

Cultural Control. Drain or empty containers of standing water. Remove old tires. Drain water-holding areas around the facility. Keep waste lagoon margins free of vegetation and floating debris (Rutz and Axtell 1978).

Chemical Control. Adulticides and larvicides can be used.

Monitoring (weekly or at least once every two weeks)

Adults. Inspect the walls, ceilings, and eaves of the facility, and the vegetation around the outside of the facility for resting adults. Light traps can also be used.

Larvae. Inspect probable breeding areas such as containers of water, water-holding areas around the facility and waste lagoons. Mosquito abundance in lagoons and drainage ditches around the facility can be determined by dipping (10-20 dips) with a water dipper around the entire lagoon and/or ditch margin.

Poultry Pests: Darkling Beetles (lesser mealworms)

Management

Residual insecticide applications to posts, walls, and ceilings are effective in managing darkling beetles. Litter treatments, particularly around the posts and walls, are also effective.

Monitoring

Inspect the curtains, cracks and crevices, and the litter, particularly around the posts, walls, and feeders at least once every two weeks. Night inspections of posts and walls with a flashlight help in determining the severity of the darkling beetle problem.

Poultry Pests: Rodents (Norway rat (common names: brown rat, house rat, barn rat, sewer rat, and wharf rat), roof rat, house mouse)

Management

Cultural Control. Rodent proof and eliminate harborages. Keep vegetation cut around the facility. Raise and lower the side curtains at least once a week during the warm months when they are not in use.

Chemical Control. Anticoagulants (multiple dose) and acute rodenticides (single dose) can be used. Bait placement is very important. Bait should be placed throughout the inside and outside of the facility and as close to active burrows as possible, preferably within the burrows.

Monitoring

Inspect both the inside (egg rooms, along side walls, under

slats, etc.) and outside of the facility for live rodents, droppings, and active burrows at least once every two weeks. To determine if a burrow is active, fill it in with dirt or litter. An active burrow will be opened up again in two or three days. Inspect curtains, electrical wiring, and insulation for rodent damage.

Poultry Pests: Birds (chimney swifts, swallows, sparrows, and starlings)

Management

Exclude by blocking observed access holes in the facility.

Monitoring

Inspect the facility for adults, droppings, nests, and insulation damage at least once every two weeks.

Facility Inspection

Inspect each house for leaky waterers and feed systems, improperly working fans, drainage problems, and any other existing problems.

Swine Pests: External Parasites (hog louse, mange mite)

Management

New boars, sows, and feeder pigs added to a herd may carry mites and lice or their eggs to animals already present. New additions to a herd should be quarantined and treated twice (10-14 days apart) before coming in contact with the existing herd. In the existing herd, routine preventive treatment of suspected louse and mite infestations is NOT recommended. Treatment for lice should be done ONLY when lice are observed on the animals. Mite treatment should be done ONLY when suspected infestations are verified by scrapings.

Monitoring (at least once every two weeks)

Lice. Inspect the ears, skin fold areas behind the ears, neck, and back of a systematic sample (10 percent) of the animals in the herd.

Mites. Inspect the areas around the eyes, ears, neck, back, and hind legs of a systematic sample (10 percent) of animals in the herd. Scrapings must be used to verify mite infestations.

Swine Pests: Filth Flies (house fly, lesser house fly, black garbage fly, soldier fly, blow fly, flesh fly, and false stable fly)

Management

Cultural Control. Manure management - As appropriate to house type, dispose of animal wastes and other organic debris frequently (every 5-7 days). Provide proper grading and drainage around the facility. Keep grass and weeds cut to provide good air circulation to facilitate drying of manure and animal bedding.

Sanitation - Remove and properly dispose of dead animals, wet hay, straw, spoiled feed, or any other organic material that can attract and provide breeding sites for flies.

Chemical Control. A combination of adulticides and larvicides can be used. Adulticides include baits, residual sprays on upper inside surfaces where flies rest, and/or space or mist sprays. Larvicides should be used ONLY for spot treatment in those areas of the manure where large numbers of fly larvae are observed.

Monitoring

Adult Flies. Baited traps (see fly monitoring in Poultry Pest section) are used to monitor adult flies. The number of traps will vary according to facility size, but a minimum of three at equidistant locations throughout each house are used. The traps are left for 7 or 14 days, depending on the monitoring schedule, and then the number and species of flies collected are counted.

Treatment with adulticides is recommended when an average of 350 flies is caught per trap per week (700 flies per trap per two weeks).

Larvae. Inspect the manure and other potential fly breeding areas in and around each house. Spot treatment with larvicides is recommended ONLY on those areas where large concentrations of filth fly larvae are observed.

Swine Pests: Horse Flies and Deer Flies

Management

These insects are strong fliers and can move long distances from their breeding areas, making control very difficult. Residual insecticide applications to animals and buildings provide some control. Space or mist sprays can also be used.

Monitoring (weekly or at least once every two weeks)

Inspect animals for feeding adult flies. Malaise traps, Manning traps, and black panel sticky traps can also be used to monitor these flies.

Swine Pests: Stable Flies

Management

Eliminate breeding sites (wet hay, straw, manure, spilled feed, and decaying vegetation). Chemical control measures include residual insecticide applications to animals and building surfaces and space or mist sprays.

Monitoring

Inspect animals for feeding adult flies. Sticky translucent panels (traps) can also be used to monitor adult stable flies. Inspect probable breeding areas (wet straw, manure, spilled feed, decaying vegetation) for stable fly larvae.

Swine Pests: Mosquitoes (see Mosquitoes in Poultry Pest section)

Swine Pests: Rodents (Norway rat (common names: brown rat, house rat, barn rat, sewer rat, and wharf rat, roof rat, house mouse) (see Rodents in Poultry Pest section)

Facility Inspection

Inspect each house for leaky waterers and feed systems, improperly working fans, drainage problems, and any other existing problems.

ECONOMIC THRESHOLDS

The economic thresholds used in the program are guidelines used by the agent in determining recommendations. Facility type, time of year, age of animals, pest population change over time, and proximity to housing developments are all taken into consideration before economic thresholds are determined for a particular facility. These values will be continuously updated as the program progresses.

PRODUCER RESPONSIBILITIES

1. Provide access to animals and facilities.
2. Maintain a written record of pest management activities, such as insecticide applications (insecticides and amount used), rodent baiting, etc.
3. Share in program costs (fees).

PROGRAM FEES

Poultry Facilities

Caged-layer, broiler-breeder, broiler, turkey breeder and turkey fryer. The base fee is $4.00 per visit plus a 10¢/1,000 bird head fee. Example: The fee for a 25,000-bird facility is $6.50 per visit ($4.00 base fee + $2.50 head fee).

Swine Facilities

Breeder facilities - Base fee $4.00 per visit plus a 3¢/sow head fee. Example: The fee for a 300-sow facility is $13.00 per visit ($4.00 base fee + $9.00 head fee).

Feeder facilities - Base fee $4.00 per visit plus a 25¢/100 animal head fee.

The treasurer's office in each county bills cooperating producers and/or their integrators quarterly (every three months). Some integrators pay the program fee for their producers, others pay half and the producer pays half of the fee, and some producers pay the entire fee themselves.

TION

rict sanitation to prevent diseases from being carried from one production facility to another is mandatory while working with poultry and livestock. The sanitation procedures used by the program pest management agents include the following:

1. Agent vehicles are parked at a reasonable distance from the facility.
2. Sanitized boots and coveralls are worn by the agent at all times while performing his duties at each facility.
3. The agent, upon arrival at each farm, puts on a clean pair of coveralls, and scrubs and rinses all boots, ladders, and other equipment with disinfectant before entering the facilities.
4. After the agent has completed his duties, coveralls are removed and placed in a laundry bag, and all boots, ladders, and other equipment are again scrubbed and rinsed with disinfectant.
5. Similar types of facilities are visited on the same day. Caged-layer facilities are visited on one day, broiler-breeder facilities on another day, etc. There is NO agent movement between different facility types (caged-layer to broiler-breeder or broiler to broiler-breeder) during the same work day.

FACILITY DIAGRAMMING

A diagram of the general facility layout and a detailed floor plan of each animal housing unit is made by the agent when the producer joins the IPM program. This diagram is updated when any significant modifications are made to the facility. These diagrams facilitate communication of pest management recommendations between the producer and the IPM personnel.

POULTRY AND LIVESTOCK IPM ADVISORY COMMITTEE

The purpose of this committee is to coordinate program progress, identify areas of need, apply resources to need, and provide expertise. This committee functions in an advisory capacity to the administration of North Carolina State University and includes representatives from the Departments of Poultry Science, Animal Science, Biological and Agricultural Engineering, Economics and Entomology.

SUMMARY

The North Carolina Pilot Poultry and Livestock Integrated Multipest Management Program is an extension demonstration program designed to provide producers, farm managers, integrators, integrator field men, and county agents with information and training in the principles and application of IPM. This program involves a systems approach to pest management with poultry scientists, animal scientists, biological and agricultural engineers, economists, veterinarians, and entomologists involved in program development, implementation, and evaluation.

The program is directed at the management of the entire arthropod, rodent, and bird pest complex at poultry and livestock facilities. Caged-layer, broiler-breeder, broiler, turkey breeder, turkey fryer, swine breeder, and swine feeder producers and integrators are included on the program.

Since its inception, the program has undergone considerable change. Monitoring schedules at most facilities have changed from weekly to once every two weeks. During the first year, the pest management agent visited facilities, monitored pests, and provided recommendations. This procedure provided producers with information and training in IPM; however, the integrator field men often did not get the full benefit of this information and training. This year the agent will work directly with integrator field men by accompanying them on their farm visits and providing on-the-job pest management training for both field men and producers. In addition, IPM workshops will be conducted for producers, integrators, integrator field men, farm managers, and county agents. The inclu-

sion of internal parasite monitoring into the program, at the request of both integrators and producers, could lead to the development of a whole flock or herd health program, and is being investigated. Development and implementation of this IPM program is a continuous interdisciplinary research and extension process because of continual changes in poultry and livestock production facilities, practices, and technology, and advances in pest management technology.

REFERENCES

Anon.
1979. Proceedings of a workshop on livestock pest management: To assess national research and extension needs for integrated pest management of insects, ticks and mites affecting livestock and poultry. 332 p. Kansas State University, Manhattan.

Rutz, D. A., and Axtell, R. C.
1978. Factors affecting production of the mosquito Culex quinquefasciatus (=fatigans) from anaerobic animal waste lagoons. 32 p. North Carolina Agricultural Experiment Station Technical Bulletin 256.
1980. House fly (Musca domestica) parasites (Hymenoptera: Pteromalidae) associated with poultry manure in North Carolina. Environ. Entomol. 9: 175-180.
1981. House fly (Musca domestica) control in broiler-breeder poultry houses by pupal parasites (Hymenoptera: Pteromalidae): Indigenous parasite species and releases of Muscidifurax raptor. Environ. Entomol. (in press).

30 YEARS--AN EDUCATION IN FILTH FLY CONTROL

Edmond C. Loomis

University of California, Davis

The fundamentals of filth fly control were presented in 1948 at a conference of public health and mosquito control entomologists and vector control specialists representing local and state governmental agencies (Rowe 1948). Three basic factors were considered important when considering a fly control program: 1) education, 2) preventative measures, and 3) desire of people to have a program. By 1950 the California Department of Public Health organized a statewide fly control program directed by a sanitation engineer and an entomologist. In cooperation with local health agencies the primary targets were the conversion of garbage dumps into sanitary land fills and the development of corrective measures to minimize backyard fly development sources. Progress reports and summaries of successful operations on refuse disposal were made by Black (1954, 1958, 1964). Concurrent to this statewide effort was the role of agriculture concerned with the wastes of animal and crop production in both rural and urban areas.

The rapid increase of the human population in California following World War II initiated the eventual change from sharing 1,000 acres to only 5 acres per capita. Prime agricultural land was gradually blacktopped to accomodate the growth of urban and industrial developments, all of which brought about problems in waste disposal. Likewise, farm management practices were undergoing dynamic changes--from floor to wire-cage operations on poultry ranches, from pasture to drylot operations on dairies and a greater concentration of cattle feedlots to accomodate beef marketing centers, and from warehouse to field processing and packaging of vegetables. These are only a few examples of the demand to produce more food, a greater variety of food, and special packaging and preparation of food to satisfy a prosperous and affluent society. Thus, the core of the fly problem was concerned with wastes from living and from making a living.

The current animal and human population densities and distribution patterns in California are no different in 1980 than those

which showed directional growth 30 years ago. Of the 23.5 million people in the state, 95 percent (22 million) live in three main regions--the Central Coast, the San Joaquin Valley, and the Southern Region (figure 1). Poultry layer ranches (figure 2) and drylot dairies (figure 3) are distributed between north, central and south coastal areas as well as in the San Joaquin Valley, while beef, although statewide, occur mainly in the San Joaquin Valley, the south coast, and in two widely separated areas of the southern region (figure 4).

Fly problems and methods for control were identified in early surveys of poultry layer ranches by Smith (1954) and Hart (1957), or dairies (Murray and Whitten 1955, Smith 1956), and for farm and garden wastes (Golucke 1955). An integrated approach to waste management was necessary largely because of differences in climate and secondarily by the principle that each ranch is different. With food animal operations these differences could include one or more factors such as animal housing, ranch design, animal breeds and behavior, animal rations, formulations and methods of feeding, soil profiles, and the type and amount of cultivated acreage for fertilizer application. The last factor played a minor role prior to 1970 because commercial fertilizer was cheaper and easier to use than animal manures. The energy crisis in the 1970's reversed this situation and animal confinement operations received high prices for manure fertilizer. This then, was in contrast to the earlier years when manures were a debit and manure haulers had to be hired for animal waste collection and disposal.

In cooperation with state and local governmental agencies, industry, and food commodity associations, the University adopted an Agricultural Sanitation Program coordinated through Cooperative Extension (Loomis 1964). Four main categories for waste management included physical, mechanical, chemical and biological control. The program was supported by basic research conducted in the Experiment Stations, by field investigations and demonstrations, and by educational activities. These efforts were not the sole responsibility of any one group but rather the interrelated activities of scientists and specialists representing agricultural engineering, agronomy, animal and avian sciences, economics, entomology, range, soil, and irrigation sciences. An early success in education was a series of agricultural-public health training courses. Local representatives from major animal and crop commodities, boards of supervisors, planning commissions, public works, public health, and the University, met for 2-3 days to better understand each others problems and to develop a cooperative approach to problem solving. Besides filth fly control were those questions on the abatement of odors, dusts, feather-blowing, and even noise. One benefit derived from these courses was the integrated management of people (IMP) in cooperating on local nuisance (fly) control committees. Responsibilities included formulation of flexible ordinances, agreeable animal structure--construction

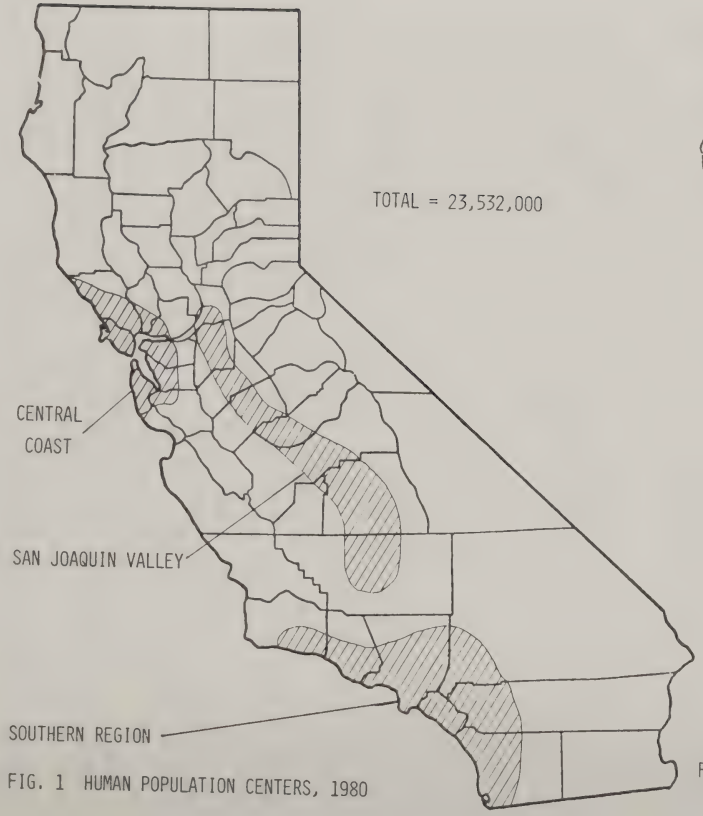


FIG. 1 HUMAN POPULATION CENTERS, 1980

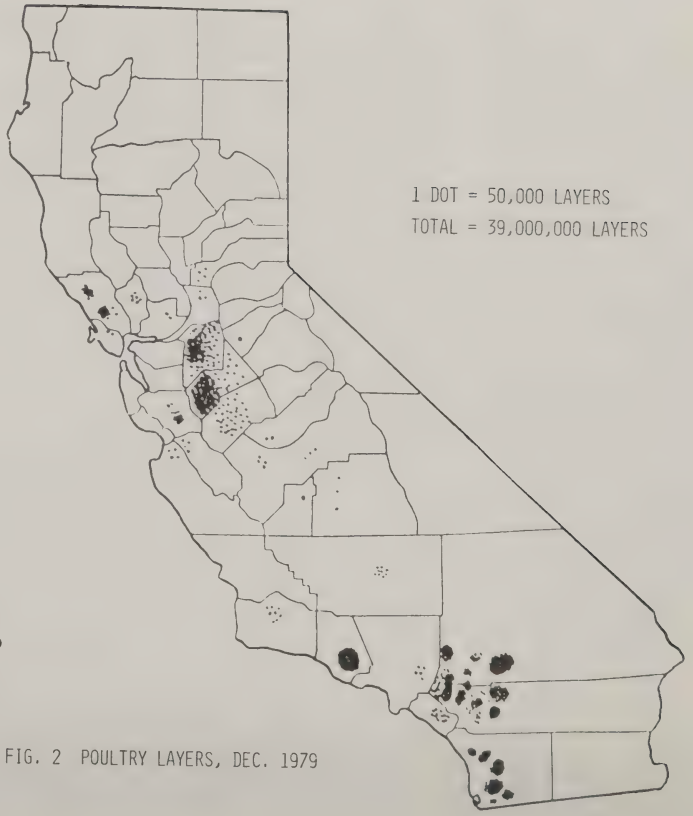


FIG. 2 POULTRY LAYERS, DEC. 1979

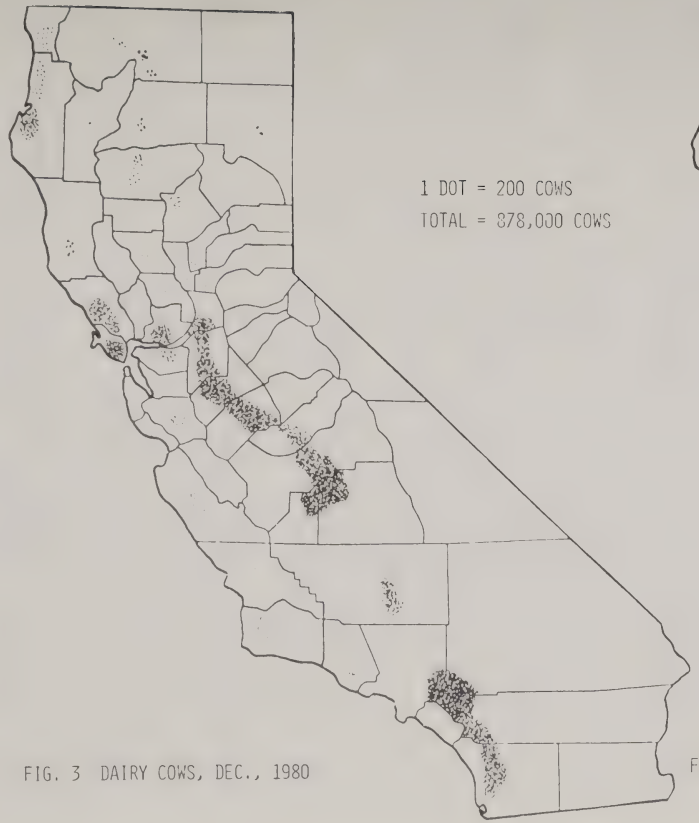


FIG. 3 DAIRY COWS, DEC., 1980

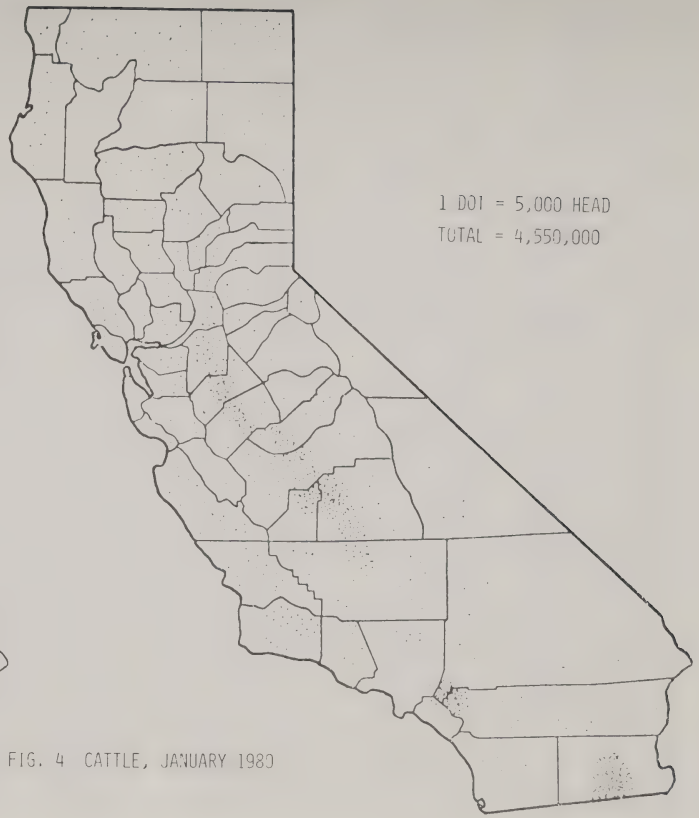


FIG. 4 CATTLE, JANUARY 1980

permits, and guidelines for use and disposal of animal and crop wastes. Action by these committees greatly reduced the need for cease and desist orders and resultant court hearings. After all, court cases which lasted for weeks to months merely permitted greater intensity of nuisance problems back on the ranch and strict ordinances in state and local public health codes never resulted in legislating flies into extinction.

The University's Agricultural Sanitation Program, therefore, assumed responsibility for "FOD" nuisance problems (flies, dusts and odors). On certain beef feedlots, however, nuisance problems were defined as "DOF" (dust, odor, and flies). Evening activity of steers in feedlot pens often created dust clouds which later moved into nearby towns with eventual complaints including those against odors. Studies were conducted to control dust and odors using optimum animal stocking rates of pens without increasing the problem of filth fly production (Miller, 1962). On poultry layer ranches, problems were minimized by seasonal timing of manure collection and disposal, and house enclosure.

The phrase "wet it or dry it" has led to the use of manure lagoons (ponds) for aerobic destruction of organic wastes. Acceptable operation of most ponds in use by dairies and poultry ranches is that where the effluent can be mixed with irrigation water to fertilize adjacent cropland. Land-poor operations involve a pump return system for wash down of cement work alleys (dairies) or under-cage cement dropping areas (poultry). Odors originating from these ponds have varied from none to objectionable, the latter arising from an overload of solids beyond the original planned pond capacity. Some liquid systems on flush clean dairies have resulted in 100 percent use of concrete-floored pens and walkways. Deodorant chemicals applied during corral manure cleanout or the rapid drying of manure for use as cow bedding in open-sided loafing barns are additional methods used by drylot dairies. Although some of these methods require a large capital investment in equipment and construction, the results of good "FOD" control have led to a greater incentive to produce a more suitable waste product for fertilizer use. These and other examples on the control of "FOD" nuisance problems are described by Loomis (1973).

Insecticide control of flies has largely been through the use of adulticides to farm structures or to animal manures as larvicides. Residual surface sprays have been effective when properly chosen and carefully applied. Most animal confinement operations, however, do not practice early season application as recommended by De Foliart (1963) and Anderson (1966). Livestock operators usually use insecticides at a time when their premises are under attack by dense numbers of flies. Thus, the frequent use of insecticides has resulted in resistance to chemicals by numerous fly pests. One activity included in chemical control studies in California was the monitoring of fly

resistance to insecticides (Georghiou and Bowen 1966). Results of this continuing activity has guided recommendations for insecticide use in critical areas and on certain livestock and poultry operations. Also, larviciding methods have been largely discontinued because of the acceleration of chemical resistance by flies and because of high mortality to beneficial arthropods which help degrade the substrate and which may act on predators and parasites of fly eggs, larvae, and pupae. Insecticide use, therefore, has decreased during the last 10 years. This is particularly true on poultry layer ranches in Southern California where the use of fly predator/parasite agents was added to the integrated control approach and more judicious use of insecticides resulted.

Investigations into biological control methods of filth flies was started during the 1960's and preliminary results were published by Legner and Brydon (1966) and by Legner and Olton (1968). Extensive studies made on poultry layer ranches during these early years demonstrated potential reduction of filth fly populations only when a general understanding was obtained on the biologies of flies and their natural enemies (Peck 1968, 1969, Legner and Olton 1971). Manure-inhabiting beetles belonging to five families (Anthocoridae, Histeridae, Scarabaedidae, Staphylinidae, Tenebrionidae) were found to contain predatory and scavenger species of filth fly eggs and first instar larvae. Also, native parasitic hymenopteran species of the genus *Spalangia* and *Muscidifurax raptor* were found to attack larvae and pupae of the house fly and the *Fannia* group. To augment these indigenous natural control agents, parasitic fly enemies were introduced to the California program of which the most effective were: three reproductively isolated forms of *M. raptor* from South and Central America and Puerto Rico, and *Tachinaephagus zealandicus* from Australia and New Zealand. The early releases of these parasitic insects on poultry ranches yielded partial success for fly control during summer (Legner and Dietrick 1972, 1974) but only fair reduction of house flies from winter releases (Olton and Legner 1975).

The results of these biocontrol studies were included in poultry ranch fly control recommendations which also stressed other measures that helped maintain fly predators and parasites (Loomis et al 1975). For example, a concrete pad beneath caged layers prevented house flies from pupating in the soil and more fly pupae were present at the dry edges of droppings where parasites are best able to destroy them; removal of the top portion of manure to leave an 8 inch pad or removal of all manure on an alternate row basis, aided in preserving the habitat of existing natural predators and indigenous and introduced parasites. These procedures, however, apply only under conditions of good manure management, including excellent water control, soft shell egg and dead bird disposal. In more recent years the commercial production of wasp parasites by the Vitova Company in California for release on poultry ranches has

assisted the overall goal of filth fly control. It is unfortunate, however, that advertizing brochures of other commercial distributors of these natural enemies, contain misleading statements as to the true efficacy and correct use of such parasitoids. Statements such as "these useful insects attack all common dung-breeding flies including the biting stable fly, housefly, horn fly, face fly, and *Fannia* species" or, "they also attack other problem flies--fruit fly, blow flies, horse flies, and flesh flies", are largely untrue and such misinformation should be corrected and policed by the industry.

In conclusion, we have solutions for the control of filth flies even beyond the historic barnyard methods of manure management and of manure use as a fertilizer. What is constantly needed is the correct emphasis and selection of one or more of the 4 basis factors--physical, mechanical, chemical, and biological, to control fly nuisance problems on individual ranches. Achievement of these goals in the agricultural sector will be compatible with program actions conducted by industry and public health agencies for community-wide fly and nuisance control.

REFERENCES

Anderson, J. R.
1966. Recent developments in the control of some arthropods of public health and veterinary importance. Bul. Entomol. Soc. Amer. 12: 342-348.

Black, R. J.
1954. Choosing a refuse disposal method. Calif. Vector Views 1: 1-3.
1958. Sanitary landfills servicing California communities in 1957. Ibid. 5: 31-32.
1964. The solid waste problem in metropolitan areas. Ibid. 11: 51-55.

Bohart, R. M.; Davis, C. S.; Deal, A. S.; Furman, D. P.; March, R. B.: Smith, A. C.: and Swift, J. E.
1958. Insecticides for control of flies on poultry ranches, dairies, homes and food processing plants. Calif. Vector Views. 5: 35-41.

De Foliart, G. R.
1963. Preventive spraying schedules for dairy farm fly control. J. Econ. Entomol. 56: 649-654.

Georghiou, G. P., and Bowen, W. R.
1966. An analysis of house fly resistance to insecticides in California. J. Econ. Entomol. 59: 204-214.

Golucke, C. G.
1955. Composting farm and garden wastes. Calif. Vector Views. 2: 58-64.

Hart, S. A.
1957. Chicken house fly control through manure handling. Calif. Vector Views. 4: 67-70.

Legner, E. F., and Brydon, H. W.

1966. Suppression of dung-inhabiting fly populations by pupal parasites. Ann. Entomol. Soc. Amer. 59: 638-651.

Legner, E. F., and Dietrick, E. I.
1972. Inundation with parasitic insects to control filth breeding flies in California. Proc. Calif. Mosq. Control Assoc., Inc. 40: 129-130.
1974. Effectiveness of supervised control practices in lowering population densities of synanthropic flies on poultry ranches. Entomophaga. 19: 467-478.

Legner, E. F., and Olton, G. A.
1968. The biological method and integrated control of house and stable flies in California. Calif. Agric. 22: 1-4.
1971. Distribution and relative abundance of dipterous pupae and their parasitoids in accumulations of domestic animal manure in the Southwestern United States. Hilgardia. 40: 505-535.

Loomis, E. C.
1964. Agricultural sanitation and the domestic fly problem. Proc. Calif. Mosq. Control Assn.32: 34-36.
1973. Agricultural sanitation of livestock manures for control of flies, odors, and dusts. Jour. Milk and Food Tech. 36: 57-63.

Loomis, E. C.; Anderson, J. R.; Bowen, W. R.; Deal, A. S.; Georghiou, G. P.; Legner, E. F.; and Swanson, M. H.
1975. Fly control on poultry ranches. Univ. Calif. Div. Agric. Sci. Leaflet 2505; 17pp.

Miller, R. F.
1962. Space requirements and dust control for feedlot cattle. Calif. Agric. 16: 13-15.

Murray, W. D., and Whitten, G. R.
1955. Mosquitoes and fly source reduction in liquid manure systems. Calif. Vector Views. 2: 11-15.

Olton, G. S., and Legner, E. F.
1975. Winter inoculative releases of parasitoids to reduce house flies in poultry manure. J. Econ. Entomol. 68: 35-38.

Peck, J. H.
1968. The potential role of arthropod predators in the integrated control of Diptera developing in poultry droppings. Ph.D. thesis, University of California, Berkeley, California.
1969. Arthropod predators of immature Diptera developing in poultry droppings in northern California. Part II Laboratory studies on feeding behavior and predation potential of selected species. J. Med. Entomol. 6: 168-171.

Smith, A. C.
1954. Flies, Chickens, and people. Calif. Vector Views. 1: 1-3.
1956. Fly prevention in dairy operations. Calif. Vector Views. 3: 57-60.

MONITORING HOUSE FLY POPULATIONS AND CARRYING OUT AN INSECT PEST MANAGEMENT PROGRAM IN CAGED LAYER OPERATIONS

Maxcy P. Nolan, Jr.

University of Georgia, Athens

Fairly recently, within the last 10 years, house fly control problems have increased with the advent of placing animals in confinement-type production facilities. Before 1970, in most areas, confinement-type production units were isolated from suburban areas. Also, most animal production units did not concentrate animals in densely populated animal housing units. Additionally, people who lived in rural areas were more conditioned to and, therefore, were more tolerant of "farm-type insects and odors." Today, the very existence of confinement-type animal operations is being threatened due to a general lack of tolerance to flies and odor by people living near confined animal operations--such as dairies, poultry facilities, swine facilities, etc.

In most confinement-type animal facilities, especially poultry facilities, it is generally accepted that house fly control is a public relations effort, since house fly control will not normally result in increased animal production efficiency unless house fly numbers become exceedingly heavy. Confinement-type animal units were designed for the efficient production of animal products and little planning was given to the management of manure to prevent house flies and odor. In many existing facilities house fly control, which results when animal wastes are managed properly, may not be achieved without major manure management modifications. As manure becomes more valuable as a source of animal feed, energy, plant food, etc., manure management practices, which result in house fly and odor control, will become more cost effective.

LIVING WITH YOUR NEIGHBOR--CONTROL FLIES AND ODOR

Americans appear to be more aware today than ever before of environmental pollution. Also, they seem to be less tolerant of nuisance factors, such as flies and odor. And to cap it off,

people who were raised in non-farm and non-rural environments are moving to farming and rural areas. They bring with them the false impression that farming areas are always free from dust and insects and that they always smell like freshly mown hay.

In many areas farmers are outnumbered and, of course, they are outnumbered nationwide. Therefore, farmers must attempt to be good neighbors. They should assure their neighbors that they are sensitive to their concerns. This genuine concern is especially important when it has an impact on farming operations and, thus, the very livelihood of the farmers.

Overall the poultry industry is a highly positive industry. For example, the broiler, turkey, and hen producers can place animal protein on the tables of all Americans at a reasonable cost. America is ready for more variety at fast food establishments; thus, chicken sandwiches and fried chicken sales are skyrocketing. Eggs are both highly nutritious and one of the best buys on the shopper's food list today. Thus, the per capita consumption of poultry meat and eggs is on the increase.

The poultry industry is an extremely valuable industry. Each and every day, in Georgia alone, approxiamtely 2 million dollars are generated at the farm level by the poultry industry. This figure is multiplied many times when we take into consideration the total impact the industry has on Georgia's overall economy and that of other poultry producing states.

Positives, positives, positives. With so many positives stacking up on the side of the poultry industry and on the side of producing poultry meat and eggs, then how could there possibly be any negatives? Poultry farmers should be assured that there are negatives which can impact directly on the poultry industry. These negatives must be identified and dealt with in a way that will be economically feasible. Flies and odor which can be produced in poultry operations can be negative factors that could impact unfavorably on community relations.

Therefore, producers should first determine if flies and odor are being produced in their poultry operations. If they are, then steps should be taken to correct the problem and prevent the spread of these problems to their neighbors.

The best time to handle a fly and odor problem is before a problem exists. Before building a poultry operation, potential producers should seriously consider manure management alternatives so fly and odor problems will not occur. It seems to be easier to keep a happy neighbor happy than to make an angry neighbor happy--so be a good neighbor.

Why are flies and odor present around poultry operations? Why are they prooduced there? Flies and odor can be produced in and by the waste products which are being generated by the poultry housed in poultry operations. If not handled properly, poultry manure accumulations can produce excessive flies and odor.

Granted, poultry operations are attractive to flies, since adequate food, shelter and breeding sites are present for them. This makes it even more important to be a good neighbor. Cooperation will allow potential fly breeding areas in the community to be properly managed so they do not breed flies. This will help everyone. If everyone cooperates, fly and odor problems can be identified and corrected throughout the community. Everyone must do their part because fly and odor problems have always been present in the farming and urban environment; and it is not reasonable to expect one segment of the farming or urban community to shoulder the responsibility alone. Mostly, poultry producers are conscientious and work hard to correct these problems; however, their neighbors have horses, cattle, garbage containers, and other sources of flies and odor on their property and do little about the flies and odor. In these areas, poultry producers have cleaned up their problem, but as soon as they have, neighbors send them their flies and odor. In these instances, being a good neighbor and working together can pay off.

Overall, poultry manure is a valuable commodity, when it is used as animal feed, fertilizer, or a source of fuel. When used as fertilizer, poultry manure will supply plant nutrients worth $25-30 per ton ($.0125/lb. to $.015/lb.). Each commercial layer produces 40 lbs. of manure per year for a total value of $.50 to $.60. Each broiler produces 3 lbs. of manure at a value of $.04 to $.05. Broiler breeders produce 44 lbs. of manure each year at a value of $.55 to $.66, while turkeys produce 31 lbs. of manure for a value of $.39 to $.47. These values are considered very conservative.

This highly valuable commodity can become a problem. In fact, it can jeopardize the very existence of a poultry operation. If this valuable product is allowed to produce excessive flies and odor, it could cause legal problems if a nuisance is created; or it could strain relations with your neighbor. In fact, in some areas the problem has become so severe that producers have been taken to court, while others have been forced out of business. In some areas, citizens will not allow poultry houses even to be built, because they are afraid of the potential fly and odor problems. In other areas, the fly and odor problems have become so severe that the value of the industry in providing jobs and cash flow in the community has been forgotten.

It might be mentioned here that some communities have considered zoning as a means of handling this type of problem. This would be a good solution where moderate fly and odor problems were being encountered or were expected. However, it may not be the answer for some areas where fly and odor problems are massive. Then, zoning or not, the citizens in the area may get to the point that they can't tolerate the excessive flies and odor, and a court battle may ensue.

If poultry manure is removed often, diluted with water, plowed into the soil, fed to livestork, or otherwise handled so it does not accumulate, then flies and odor should not be a problem. Can a poultry producer afford to remove manure on a frequent basis? What about the expense of a flushing system? What about provisions for inclement weather that may not allow frequent disposal of solid manure on fields with spreaders? In most operations it may not be feasible or practical to remove poultry manure often and other arrangements may be needed in those facilities. Because most poultry operations will produce excessive flies and odor if the manure is not handled properly, present and potential poultry producers should ask themselves these questions, and others, to determine solutions so that these fly and odor problems do not occur.

Fly and odor problems seem to be closely dependent on the type of poultry operation. Egg producing operations include floor layers that produce table and hatching eggs, layers on 1/3 scratch and 2/3 wire or slats where hatching eggs are produced, and caged layers where table eggs are produced. The old type floor operations seldom produce flies because the poultry have access to all areas in the house. They scratch up and consume fly larvae that are produced in the litter. In most instances, the litter is too dry in floor houses to produce flies and odor. In these houses manure is removed about once each year.

Most floor operations have been remodeled into areas that consist of 2/3 wire or slats and 1/3 scratch area. Poultry droppings are deposited through the slats or wire and accumulate on the ground. These operations can produce flies and odor if the litter is not maintained in a highly dry state. Problems with waterer leaks will sometimes lead to fly and odor problems. Special care must be exercised in these houses to keep the litter dry because it cannot be removed until the birds are removed, which occurs about once each year.

Caged layer operations can produce excessive fly populations and odor problems. Here manure accumulates under layers and flies can produce in the exposed litter. In these houses it is difficult to keep the litter dry due to blowing rains, leaking waterers and loose birds. Most producers do not allow

poultry droppings to become wet (soupy) because of the above problems. If the litter is removed twice weekly, or on a more frequent schedule, and spread thinly on fields to dry or plowed into the soil, then fly problems will not occur. Of course, transporting litter could pose a problem, if it is allowed to drop on highways and other areas frequented by the public. If we cannot dispose of litter twice each week or more often, then it should kept very dry, very wet, or treated with chemicals to prevent fly and odor problems.

Poultry operations produce valuable commodities such as eggs, meat, and manure, which can be valuable when used as fertilizer, animal feed, and fuel. Thus, the poultry industry is a highly valuable one. Therefore, every feasible effort should be made to control the flies and odor produced in these operations. Potential producers are advised to get the facts even before building. When fly and odor problems occur, producers should carefully consider the alternatives in an attempt to eliminate the problems. The best information should be considered before deciding on a course of action.

CONTROLLING HOUSE FLIES IN POULTRY OPERATIONS

Learning to live in harmony with your neighbor can be very important. In fact, it may even determine the future success and continuation of your poultry business.

Excessive fly numbers can build up in a poultry operation and eventually "spill over" into surrounding areas. Adult house flies can travel long distances. They often migrate from 1 to 3 miles from their breeding site in search of food, shelter and new breeding sites. Therefore, living in harmony with your neighbor might involve fly control.

House flies breed in accumulations of poultry droppings. Each pound of poultry manure can produce from 100 to 1000 house flies. Since each layer will deposit appromixately 40 to 50 lbs. of manure each year, each has the potential of depositing suitable breeding sites for large numbers of flies. Therefore, this highly valuable commodity, poultry manure, with a value of $25 to $30 per ton (the value exceeds $40,000,000 annually in Georgia alone), can become a negative factor when it produces excessive flies.

Manure Management Directly Affects Fly Production

Dry manure, containing less than 25% moisture, will not support house fly populations. Dry manure is found in broiler operations and floor layer operations. Here the litter is not only too dry to breed flies, but birds in direct contact with litter will scratch out and consume house fly immatures.

Breeder and caged layer houses are normally constructed so that birds are not in contact with their droppings; instead, they are suspended in cages or on wire or slats above their droppings. In these houses leaking waterers, loose birds, and blowing rains normally keep droppings with sufficient moisture (26-79%) to allow fly production. Where waterers are closely maintained to prevent dripping, where roof overhangs are sufficient to keep rains out, and where birds are kept in good health and cool, the droppings under these conditions may support only very low populations of flies. Normally, however, problems do occur that are often beyond the immediate control of the producer and droppings become wet and fly populations increase. Therefore, where careful manure management will prevent heavy fly breeding in breeder houses the same degree of care may not be as successful in caged layer operations.

Liquid manure, manure with a moisture content above 80%, will not breed house flies. Except where flushing systems are installed, it is sometimes not feasible to allow manure to become liquid. It flows over walkways and is difficult to handle with tractor blades and manure spreaders that were designed for distributing solid manure. In some areas, due to fly and odor problems, caged layer operations are being built, or existing operations are being modified, to allow flushing systems to be used to handle manure cleanout; and pits or lagoons are being used to store the flushed materials. Flushing systems cost approximately $.35/bird to install and many do not allow the use of the manure for fertilizer. Of course, house flies do not complete development where flushing systems are used.

It should be pointed out that house flies have many natural enemies such as predaceous mites, parasitic wasps, and competitive flies, as well as, other organisms which serve to help the poultry producer keep fly populations in check. Predaceous mites and parasitic wasps seem to be more effective in reducing fly populations in litter which has a moisture content below 50%. Soldier fly larvae tend to overcompete for breeding areas with house fly larvae in litter which exceeds 50% moisture content. Where practical, manure should be managed in such a way that the benefits derived from "beneficials" would be enhanced.

As can be seen, it is very important to consider fly control (for all practical purposes this is synonymous with manure management) even before a poultry operation is built. The way the manure is managed will greatly affect fly development. In nearly all poultry operations where birds are suspended above their droppings, house fly numbers will become excessive. Therefore, plans should be made to control excessive house fly populations as they occur in poultry operations and begin migrating to surrounding areas.

The house fly passes through four stages in its development (egg, larva, pupa, adult). After mating, adult female house flies lay eggs on decomposing organic matter, such as poultry manure. About 500 eggs are laid by each house fly in 2 to 7 batches of 75 to 150 eggs per batch. The tiny (25 laid end-to-end equal 1 inch), whitish and elongate eggs hatch in 2 to 30 hours into small white larvae (maggots). The larvae tunnel into the breeding material and feed for 2 to 14 days. They molt twice during this period. Upon reaching maturity, the larvae are 1/2 inch long. They seek a dry place to pupate. The third larval skin hardens into a case called the pupal case. Inside the pupal case the larval fly transforms into a winged adult fly in 1 to 10 days. After emergence, adult flies begin laying eggs in 2 to 23 days. Adult flies will live approximately 20 to 45 days.

Upon examining each of the life stages of a house fly we can determine that the egg, larval, and pupal stages will be found in the breeding material or in the immediate vicinity (some mature larvae and pupae may be found in dry areas such as the soil near breeding areas throughout the year). Removing the breeding material and disposing of it properly will result in breaking the life cycle of the fly. Care must be taken when removing manure to remember that if manure is removed that contains mature larvae, they can tunnel into the soil in pastures to complete development even after the manure is spread. Also, adult house flies will emerge from pupal cases even though the manure is spread on fields. Spreading the manure thinly in open fields will allow immediate exposure of house fly eggs, larvae, and pupae. Most of them will be destroyed by heat, cold or other environmental factors.

If manure management does not prevent the production of excessive house fly populations in the poultry operation, other measures should be taken to control house flies and prevent their eventual build up and movement to homes, businesses, schools and surrounding areas.

House fly control programs for poultry operations should be divided into two major areas: (1) The larval house fly control program and (2) The adult house fly control program. Each program should be operated efficiently and in such a manner that each will enhance and strengthen the other. This will provide an effective, yet safe, house fly control program.

THE LARVAL HOUSE FLY CONTROL PROGRAM

Poultry manure should be checked frequently to determine if house fly larvae are present. This is especially important immediately following manure clean out. Approxiamtely 2 to 10 days following clean out examination of the newly deposited manure will reveal house fly larvae. To prevent the emergence of adult house flies we must attempt one of the following: (1)

remove the manure and spread it thinly on fields to dry, (2) bury it by plowing or subsoil injection, (3) flushing it into a lagoon or otherwise covering it with water, (4) drying it, (5) releasing a sufficient number of an effective house fly parasite, (6) by mechanical manipulation of the manure, and (7) by treating it with chemicals, etc.

Most often, immature house flies are controlled with insecticides. Often, due to labor problems, weather problems, cost effectiveness, etc. almost all fly control efforts eventaully become ineffective and insecticide applications emerge as the most dependable and often least costly method of controlling house fly larvae in manure accumulations. The dependence on larvicides does have drawbacks: (1) insecticides are required which could be costly and unsafe if not handled properly; (2) larviciding equipment is needed; (3) labor is required; (4) it may not be 100% effective; (5) this method of fly control has the potential of breeding resistance to insecticides into the fly population; (6) water is required which tends to wet the litter; (7) beneficial insects, mites, etc. are often killed along with the fly larvae. If larvicides are used, compare effectiveness, safety, cost, and availability before deciding on the insecticide to chose--and read, understand and follow all insecticide label precautions.

Recommended manure sprays include: Cygon (dimethoate) 1% spray, Rabon (Poultry Spray) 1% spray, Ravap (Poultry Spray and Larvicide) 1% spray, Residual Surface Spray and Larvicide (Supona or Compound 4072) 0.3% spray, and Vapona 0.5% spray. Applications of manure drenches should be made to thoroughly saturate the breeding material. Approximately 1 gallon of finished spray will be needed for every 25 to 100 sq. feet of manure surface. As the breeding material accumulates over 6 inches deep, then complete saturation may not be necessary if larvae are not present in the lower parts of the manure pit. Care should be taken to cover all areas of the surface of the breeding material. Poor application techniques will result in poor control. Continue to examine the manure and reapply larvicides as general surface treatments or as spot treatments only as needed. It is especially important to realize that when larval house flies transform into the pupal stage insecticide paplications are ineffective; and, normally, adult house flies will emerge from insecticide treated, as well as untreated, pupal cases.

While applying manure drenches to control house fly maggots can provide effective control, to be most effective it is necessary to incorporate an adult house fly control program. Controlling adult house flies is important because the adult stage of the fly is the migratory stage and can infest areas far from the breeding area.

Since it is virtually impossible and usually impractical to prevent all house fly production in poultry manure accumulations, producers should monitor house fly populations to determine when adult populations become excessive and begin migrating from poultry operations to surrounding areas. In studies conducted at the University of Georgia, it appears that 3 x 5 inch (7.6 x 12.7 cm) index cards can be used to monitor adult house fly populations. Relative adult house fly population density data, as measured by the number of fly specks on 3 x 5 inch white paper index cards and fly landings on Scudder grills, were used for estimating fly populations in a caged layer poultry operation. The ratios between the actual fly population and fly landings on grills (fly specks on index cards) was 1:2:5 (7.54 adult house flies per square foot is reflected as 14.8 fly specks per 3 x 5 inch index card and 34.9 fly landings/Scudder grill).

Data based on field observations indicate that excessive migrations of adult house flies from caged layer poultry operations occurs when fly specks exceed 25 specks per card per 24-hour period. Additional studies should be conducted to determine more precisely adult house fly migrations based on environmental conditions, house fly behavioral factors, etc.

THE ADULT HOUSE FLY CONTROL PROGRAM

Insecticides can be applied in a number of ways to control adult house flies. Residual surface sprays can be applied to fly resting areas. Before applying residual surface sprays look around the poultry operation at night or during a rain when adult house flies are inside the poultry operation. Most flies will be resting overhead on the ceiling, rafters, overhangs, and posts. Apply residual surface sprays to these house fly resting areas. Recommended residual surface sprays include: Atroban (permethrin), Ectiban (permethrin), Rabon (Poultry Spray), and Rava¼ (Poultry Spray and Larvicide). About 1 gallon of finished spray will be needed for each 750 to 1000 square feet to wet fly resting areas to the point of runoff.

Contact sprays can be applied in enclosed areas to control adult house flies. The purpose of these sprays is to kill adult house flies on contact. Good kill can sometimes be obtained in open areas, but best results will be achieved when the fly infested area can be closed. Insecticides recommended for contact sprays include: Dibrom (naled), pyrethrins and Vapon (dichlorvos). Up to 1 fluid ounce of finished spray may be needed for each 1000 cubic feet of infested area. Contact sprays can be applied using automatic systems, ULV foggers, misters, etc. Good coverage is necessary to completely treat the infested area before flies can escape. Where labor is a problem automatic systems are preferred.

Insecticide impregnated sweet baits can be used in certain areas and will provide some suppression of adult house flies. This method of applying insecticides can be very expensive and may not be cost effective unless used carefully. Applications to concrete aprons around egg rooms and at the end of houses where flies congregate could be effective. Since flies can become bait shy after a few months of use, producers will need to be ready to switch baits when needed.

The following is an example of a recommended house fly control program:

1. Before and after building, consider manure management alternatives to reduce fly problems.
2. Try to keep manure wet so soldier fly larvae will assist in house fly control, or keep the manure dry so predaceous mites and fly parasites will help control house flies.
3. When manure cannot be removed on at least a weekly schedule and is allowed to accumulate for periods over one week, examine the manure for fly breeding areas and be prepared to apply manure drenches of insecticides when needed.
4. Apply insecticide sweet baits in localized areas year round to control adult house flies. A small amount applied to warm sunny spots will be effective even during winter months. Heavy use of baits may not be cost effective.
5. Apply residual sprays to adult fly resting areas as needed. Permethrin seems to be especially effective at present and should be considered by all poultry producers as a part of their fly control program.
6. Contact sprays can be very effective in reducing adult house flies. Automatic systems should be considered where labor is a problem.

Exhaustive tests have been conducted and are continuously being conducted in an effort to determine the most cost effective fly control program. An effective house fly control program should be designed for each poultry operation and refinements made as needed!!! Don't be led blindly into spending money for fly control programs and techniques that will not work. Get the facts from a reliable source.

House fly control can be obtained in poultry operations. For many producers it is no longer a luxury but a necessity, since it could very well determine the very future of the industry.

INTEGRATED PEST MANAGEMENT OF FILTH BREEDING FLIES AT CAGED LAYER OPERATIONS IN FLORIDA

Philip G. Koehler

University of Florida

The most important management problem facing poultry producers in Florida is waste management and fly control. In the past twenty years the poultry industry has shifted from many small farm flocks to a few large poultry operations. Large poultry farms result in concentrated breeding areas for flies and large volumes of waste which must be successfully managed.

Florida is rapidly becoming an urbanized state with non-farm residents moving closer to agricultural enterprises. With an expanding poultry industry and urbanization of rural areas, increased demands have been placed on the poultry industry. For example, in the Tampa Bay area of Florida, there are six million chickens within 35 miles of downtown Tampa. For Tampa Bay poultry producers, waste management and fly control has been important for their industry to survive. Fly and odor control has been an important and costly public relations program which has paid off.

The management program by poultry producers around Tampa Bay was developed mainly through their own innovation. It has been extremely effective, and in 1980 not one complaint about fly production at poultry farms was brought to the county commission.

MANAGEMENT OF POULTRY WASTES IN FLORIDA

Most poultry producers in Florida manage manure to be as dry as possible. Of course, with Florida's warm, humid climate and about 60 inches of rainfall per year, dry manure is difficult to achieve. Most poultry housing is open and blowing rains in summer can often thoroughly wet manure. To overcome the adverse environment, caged layer housing should be built on an elevated grade with enough eave overhang to protect chickens and poultry waste from moisture (figure 1).

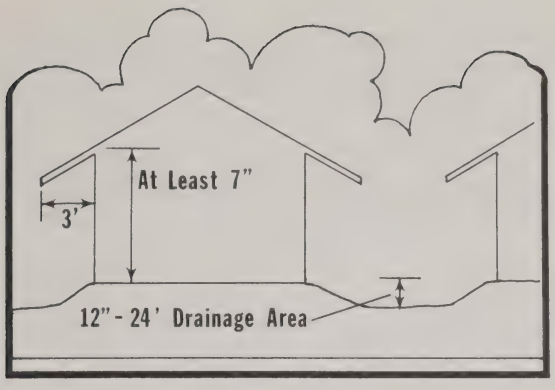


Figure 1.--Poultry house built on elevated grade.

Ventilation is necessary for dry waste management. The area around the poultry houses should be covered with grass and kept mowed. Hedges, trees, tall weeds, debris, and other materials which restrict air flow should be eliminated.

Waste removal from the poultry house is also important. The most desirable time of the year to remove waste (figure 2) is just prior to the dry months which normally occur in the fall (October and November) and spring (April and May). By removing waste before the dry months it is easier to establish

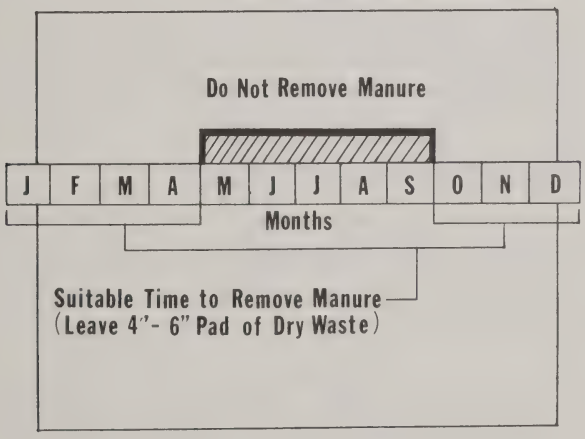


Figure 2.--Suitable times to remove manure.

dry waste under the cages. When dry waste is removed from under the cages, a dry pad of 4-6 inches of waste should be left under the cages. This aids in establishing and maintaining the drying process and natural enemies that eliminate house flies.

The hot, humid summer months of June, July, August, and September are the time when most of the heavy rains occur and this time of year is not suitable for removing waste.

The maintenance of the watering system and the roof to prevent leaks is extremely important. Leaking troughs and cups probably create most problems in attempting to keep waste dry. The watering system in a caged layer operation requires constant maintenance.

NON-CHEMICAL FLY CONTROL

Non-chemical fly control is the preferred method of fly control whenever it can be implemented. There are several reasons for non-chemical control:

Cancellation of Pesticide Registrations

Pesticides are being cancelled faster than new registrations are being granted. Therefore, fly control with pesticides is more difficult since fewer pesticides are available.

Pesticide Resistance

House flies in Florida have been found to be more than 500 times more resistance to certain pesticides than susceptible fly populations (R. Roberts, personnal communication). Therefore, it takes more pesticide to do the same job. A total reliance on pesticides required increasing quantities of chemicals and money to do an equivalent job.

Pesticide Contamination of Poultry Waste

The use of pesticides in poultry waste results in pesticide residues in waste. With increasing utilization of poultry waste as a feed additive for cattle and fertilizer for food crops, it is essential that pesticides do not find their way from waste into our food.

Protection of Beneficial Organisms in the Poultry House

Pesticides kill beneficial insects and other arthropods in the waste, as well as house flies. The use of pesticides can result in resurgence (an outbreak of house flies because the flies are resistant to pesticides and the house fly enemies are not). Non-chemical control methods are compatible with and encourage larger populations of beneficial organisms.

There are several proven methods of house fly control by non-chemical means. These are:

Waste Management

Waste management is the reliance on sanitation, weed control, proper cleanout schedules, moisture prevention, and moisture control.

Biological Control

Biological control of the house fly is a concept that has recently been developed and is currently being implemented by private industry.

Rotovation

An industry implemented practice of tilling waste to dry it.

Integrated management of filth breeding flies on Florida poultry farms has incorporated the use of waste management, biological control and rotovation in combination to reduce pest problems. Of course, the use of pesticides is minimized.

ROTOVATION OF POULTRY WASTE

Tilling waste for aeration and drying is a recent innovation in Florida. The history of rotovation began in the mid-1960's when a poultry producer named Rich Richardson developed a waste stirrer (nicknamed "rectal resource renovator") for his 94,000 bird farm with colony cages.

In 1972, another producer named Herschel Cannon built a rotovator (nicknamed "fecal fixer") and deserves a lot of credit for developing and implementing the waste management program. In 1972, he was the only producer rotovating in the Tampa Bay area of Florida. In 1977, more than 50% of the area's 200 producers had rotovators. In 1980, approximately 80% of the poultry producers were regularly using rotovators.

Rotovation works by breaking the waste into clumps, increasing surface area and causing rapid drying (table 1). Rotovation also breaks down the cone under the cages allowing better air circulation. Thus, rotovation allows better evaporation of moisture from the waste.

Hogsette (1980) analyzed manure tilled twice weekly for moisture content and numbers of house fly larvae. Tilled plots had only 40% of the numbers of house fly larvae in the manure compared to non-tilled plots. Areas not rotovated increased in moisture by 15% as opposed to decreases of over 7% for tilled plots. McKeen and Rooney (1976) have pointed out that tilling poultry manure does not guarantee dry manure. The environment and

Table 1.--Description of poultry waste after initiation of tilling two times per day (Hogsett 1980)

Time	Description of Waste
Pre-treatment	Shapeless mass with consistance of thick paste.
1 week	Consistancy of mashed potatoes and able to hold shape.
2 weeks	Manure held shape and began to break into chunks.
3 weeks	Manure broke into 3-5 cm chunks which are crusty on outside.

management programs other than tilling have a significant effect on the moisture content of poultry waste.

Rotovation is usually accomplished by attaching a commercial tilling device to a low profile diesel powered tractor. Analysis of time required for rotovation indicates that a 300 ft. poultry house can be tilled in 5-10 minutes. Hogsette (1980) found rotovation required 9.42 minutes per 91.4 meter poultry house and cost $642.58 per year or 3.2 cents per bird per year. Most pesticides are now priced at 6-8 cents per bird per year for fly control.

OVERALL FLY MANAGEMENT

The overall fly control program which was implemented by poultry producers in the Tampa Bay area of Florida was surveyed in 1977. The results of the survey are included in table 2. Poultry producers were found to be relying heavily on rotovation (55%). Those on rotovation programs were more satisfied with their fly control than producers relying on chemical control.

Since rotovation is a new control technique, many factors have to be investigated to determine why it works for so many Florida producers. Producer satisfaction indicates that it is a highly successful method of fly control. However, rotovation does require persistence and must be implemented frequently. It does not overcome poor waste and water management programs, but it does supplement them. Many poultry producers are now integrating parasite releases and rototilling to control house flies since the two methods of fly control are compatible.

Producers must realize that there are advantages and limitations to any control program. It is their responsibility to find the system which works best with their farm, watering system, waste management, and other farm practices. Carefully thoughtout programs result in excellent fly and odor control.

Table 2.--Poultry producer questionnaire Plant City, Fla. 1977

Total--29 producers responded

Size of operation
number of birds--1,358,500 = 46,845 birds/producer
(10% of total Florida layer industry)

Does watering equipment present water spillage?	
Yes	83%
No	17% (4 hart cup, 1 dew drop)
Is poultry house constructed to prevent rain from soaking waste?	
Yes	93%
No	7%
Waste removal schedule	
12 months	17%
6 months	59%
weekly	3% (scraper pit)
Fly problems	
house fly	62%
soldier fly	34%
none	14%
Type of fly control	
nothing (natural parasites)	14%
larvicide	14%
adulticide	17%
parasite release	0%
rotovation	55%
lagoon	3%
frequent removal	3%
Rotovation frequency	
as needed	6%
weekly	6%
2 times/week	37%
3 times/week	37%
4 times/week	6%
daily	6%
Chemical control frequency	
as needed	14%
monthly	14%
weekly	28%
2 times/week	28%
daily	14%

Table 2.--Poultry Producer Questionnaire--Continued

Satisfaction with fly control program	Yes	No
rotovation	94%	6%
chemical	25%	75%

REFERENCES

Hogsette, J. A.
1980. The evaluation of poultry pest management techniques in Florida poultry houses. 307 pp. Ph.D. diss., University of Florida, Gainesville.

McKeen, V. D., and Rooney, W. F.
1976. A study of fly control by stirring poultry droppings with and without a bacterial spray. Progress in poultry, Univ. Calif. Coop. Ext. Serv. Aug; 4 pp.

MASS PRODUCTION OF *SPALANGIA ENDIUS* WALKER FOR AUGMENTATIVE AND/OR INOCULATIVE FIELD RELEASES

Philip B. Morgan

U.S. Department of Agriculture

INTRODUCTION

Since house flies (*Musca domestica* L.) and other filth breeding flies, have developed resistance to insecticides, investigators have turned their attention to biological control. Here at the Insects Affecting Man and Animals Research Laboratory, over the past 8 years the microhymenopteran pupal parasite *Spalangia endius* Walker has been evaluated as a biocontrol agent against field populations of house flies. Sustained releases of *S. endius* have been successfully conducted against field populations of house flies as well as *Stomoxys calcitrans* (L.) (stable flies) at poultry, beef, dairy and swine agricultural installations (Morgan 1980b, Morgan and Patterson 1977, Morgan et al. 1975, 1976b, 1981a,b). However, a major prerequisite for successful augmentative and/or inoculative releases with *S. endius* is development of economical and efficient methods for mass culturing.

PARASITE REARING

Although *S. endius* was successfully reared by the method described by Morgan (1980a, Morgan et al. 1978) modification in these procedures have resulted in more efficient and economical methods of producing *S. endius*.

A special escape-proof Plexiglas exposure cage (61 x 61 x 48 cm) (Morgan et al. 1978) was used. The openings were covered with stainless steel cloth (70 meshes per line inch). This exposure cage was held in a room maintained at a constant 23.3°C and 70 to 80% relative humidity. Approximately 200,000 two-day-old *M. domestica* host pupae were placed in aluminum trays (ca. 33,000/tray) in each cage 4 days of each week (Monday through Thursday) and exposed to the female parasites at a preselected parasite:host ratio of 1:5 for 18 to 24 hr. The wasps dispersed throughout the pupae to oviposit and obtain nourishment from the haemolymph of the host.

After the 18 to 24 hr exposure period the majority of the parasites were separated from the pupae by attracting them to another part of the cage with either a fluorescent or incandescent light. Then the trays of pupae were emptied onto a screen (25 squares/cm^2) and the living wasps that were not attracted to the light source were sifted out by gentle agitation. This procedure increased the efficiency of the mass culturing technique by retaining in the cage a majority of the surviving female wasps. The number of female wasps was kept at a predetermined ratio by adding just enough females to compensate for the daily loss rate of 33% (Morgan et al. 1976a). When the pupae were removed another lot of 200,000 two-day-old *M. domestica* host pupae were added. *Spalangia endius* females apparently honor oviposition site markings of other *S. endius* females since superparasitism was not a problem if the 1:5 parasite:host ratio varies, or the exposure period lasts longer than 24 hr.

The pupae that were removed from the parasite exposure cage were divided into lots of 20,000 and placed in No. 8 nail bags which were then sealed. Two-thirds of the pupae were held at 27.8°C while the remaining one-third were held at 25°C. The relative humidity for both temperatures was maintained at a constant 70 to 80%. These 2 temperatures controlled the parasite larval development so that parasites that developed at 27.8°C emerged on the 21st day while those that developed at 25°C emerged on the 25th day. This guaranteed emergence of parasites Monday through Sunday with an overlap on Monday.

These temperatures and relative humidity were important. When *S. endius* was cultured at temperatures below 25.5°C, contamination by 2 other species of pupal parasites *Muscidifurax raptor* Girault and Sanders and *Pachycrepoideus vindemiae* (Rondani) often occurred. With a shorter life cycle of 13 to 18 days both species will tend to out-compete the *S. endius*. However, high temperatures affected the fecundity of both *M. raptor* and *P. vindemiae* so that the *S. endius* colony could be kept pure by culturing at 27.8°C. If the relative humidity fell to less than 60% at any time during *S. endius* larval development at either 25°C or 27.8°C, dessication occurred which resulted in death of both the parasite and the host.

Five days after the parasites were exposed to the host pupae, the adult *M. domestica* that had emerged from the nonparasitized pupae and empty puparia were removed. The remaining parasitized pupae were divided into lots of 20,000, placed in 1-liter paper containers, and held at the required temperature and relative humidity until the next generation of wasps emerged. From each batch of 100 parasitized pupae, 80 *S. endius* parasites emerged of which 53 or 66.66% were female. This sequence, when followed daily, insured the production of 1,000,000 *S. endius* per colony cage per week.

Restricting the parasite host exposure time to 18 to 24 hr allowed synchronous development at 27.8°C for a 21-day life cycle and 25°C for a 25-day life cycle of the next generation of parasites. This in turn simplified the process of obtaining adult wasps for resupplying the oviposition cage and for field releases. Out of each group of 100 parasitized pupae 53% were females. This, as well as the 33% daily loss rate, made it possible to maintain a female parasite:host exposure ratio of 1:5, which had been found to be the most efficient ratio for mass culturing the parasites.

For the field releases, the daily parasite host exposure schedule made it possible to combine parasitized pupae for a single release to ensure continuous emergence of wasps over an extended period without the need for daily travel to the release site.

REFERENCES

Morgan, P. B.
1980a. Mass culturing three species of microhymenopteran pupal parasites, Spalangia endius Walker, Muscidifurax raptor Girault and Sanders, and Pachycrepoideus vindemiae (Rondani) (Hymenoptera: Pteromalidae). VIII Renunion Nacional de Control Biologico, Tecomon, Colima, Mexico. April 22-25, 1980.
1980b. Sustained releases of Spalangia endius (Hymenoptera: Pteromalidae) for the control of Musca domestica L. and Stomoxys calcitrans (L.) (Diptera: Muscidae). J. Kans. Entomol. Soc. 53: 367-372.

Morgan, P. B.; LaBrecque, G. C.; and Patterson, R. S.
1978. Mass culturing the microhymenopteran parasite, Spalangia endius Walker. J. Med. Entomol. 14: 671-673.

Morgan, P. B., and Patterson, R. S.
1977. Sustained releases of Spalangia endius to parasitize field populations of three species of filth breeding flies. J. Econ. Entomol. 70: 450-452.
1978. Culturing microhymenopteran pupal parasitoids of muscoid flies. pp. 32-33. In Facilities for Insect Research and Production. USDA Tech. Bull. 1576.

Morgan, P. B.; Patterson, R. S.; and LaBrecque, G. C.
1976a. Host-parasitoid relationship of the house fly, Musca domestica L., and the protelean parasitoid, Spalangia endius Walker. J. Kans. Entomol. Soc. 49: 483-488.
1976b. Controlling house flies at a dairy installation by releasing a protelean parasitoid Spalangia endius Walker. J. Ga. Entomol. Soc. 11: 39-43.

Morgan, P. B.; Patterson, R. S.; LaBrecque, G. C.; Weidhaas, D. E.; and Benton, A.
1975. Suppression of a field population of house flies with Spalangia endius. Science 189: 388-389.

Morgan, P. B.; Weidhaas, D. E.; and Patterson, R. S.
1981a. Host-parasite relationship: Programmed releases of Spalangia endius Walker and Muscidifurax raptor Girault and Sanders, against estimated populations of Musca domestica L. J. Med. Entomol. 18: 158-166.
1981b. Host-parasite relationships: Augmentative releases of Spalangia endius Walker used in conjunction with population modeling to suppress field populations of Musca domestica L. J. Kans. Entomol. Soc. (In Press).

MASS PRODUCTION OF *MUSCA DOMESTICA* L.

Philip B. Morgan

U.S. Department of Agriculture

INTRODUCTION

The house fly, *Musca domestica* L., is used as a standard test animal by most state, federal and private research facilities to screen potential insecticides, rear Hymenopteran pupal parasites, and to run various physiological and biochemical studies. However, often the results of these studies cannot be replicated by these same institutions due to variation in the quality of flies used in the studies. Because house flies can survive on various substrates, many different larval and adult diets, holding rooms, and cages have been used. For the research conducted at the USDA-SEA/AR, Insects Affecting Man and Animals Research Laboratory it has been found that a consistent quality insect at an economically efficient cost can be obtained by rearing the house flies according to the methods described by Morgan (1980), Morgan and Patterson (1978), Morgan et al. (1978), and Wilson (personal communication).

EGGS

Eggs were cultured from the house fly colony by placing a small paper cup containing aged larval medium into a cage of sexually mature flies (5-6 days old). The medium was covered with black cloth to facilitate removal of the eggs. Usually an exposure of the aged larval medium of 2 hr provided an adequate supply of eggs. The eggs were washed gently from the cloth, concentrated, and then measured volumetrically in a plastic 15 ml graduated centrifuge tube. To avoid mortality the eggs were separated from the water within 5 minutes. A volume of 1 ml approximated 9,000 eggs. The eggs were placed in larval medium and held for development.

LARVAE AND PUPAE

Originally, at the Insects Affecting Man and Animals Research Laboratory, the larval medium was prepared by mixing 0.946

liters of ground oat hulls, 0.946 liters of whole oat hulls, 1.89 liters of ground alfalfa, 4.73 liters of soft wheat bran, 14 grams of commercial malt, 14 grams of brewer's dried yeast and 4.73 liters of water. The ingredients were mixed and allowed to stand for 24 hr to allow fermentation to occur. This would produce from 1,000-1,200 house fly pupae (Wilson, personal communication). Later this was replaced by a standardized larval medium consisting of 340 grams of CSMA larval medium mixed with 750 ml of an aqueous suspension containing 15 grams of moist cake yeast and 10 cc of dimalt. This mixture would produce ca. 2,000 house fly pupae (Anonymous 1958). Eventually, the yeast and dimalt were deleted from the mixture and 2.26 kg of CSMA medium moistened with an equal amount of water was the standard larval medium for many years and when 3 ml of eggs were added, 18,000 to 20,000 pupae (13-14 mg) were produced. In 1978 the price of commercially-produced CSMA medium increased from $181.00 to $725.00 per metric ton, which in turn increased the cost from $25.00 to $99.00 to produce 1,000,000 house fly pupae. This cost was reduced by diluting the CSMA larval medium with soft wheat bran (1:1) which cost $167.00/metric ton. This reduced the cost to $72.00 per 1,000,000 fly pupae. However, this combination resulted in greater compaction of the larval medium and reduced fermentation, both of which drastically affected larval development and produced small fly pupae (9 to 11 mg), which when used as a food source for parasites produced small and less vigorous females as well as erratic test results when the adult flies were stressed by exposure to chemosterilants and insect growth regulators. To reduce the cost of producing more vigorous house flies, a substitute larval medium was developed. The original CSMA larval medium consisted of 40% brewer's dried grain, 33.33% wheat bran and 26.67% alfalfa (Habermann, personal communication). All of these materials could be purchased locally with the exception of alfalfa, but pelletized coastal Bermuda, a suitable substitute was available locally. The larval medium was prepared by mixing the locally-obtained ingredients (2.266 kg) with 4 to 5 liters of water. This mixture when placed in a plastic tray (50 x 40 x 10 cm) with 3 ml of house fly eggs resulted in the production of 22,000 to 24,000 pupae weighing 15 to 18 mg. Because pupation occurred only in the top 2 to 3 cm of the larval medium, only that portion of the medium was removed. The remainder was discarded or used as oviposition medium to collect eggs. The pupae and medium were placed in a deep container (113 liters) filled almost to capacity with water. The pupae and medium that floated on the surface were gently stirred. Within a short period of time most of the medium settled to the bottom leaving clean pupae floating on the surface. The pupae were transferred by means of interconnecting pipes, to 2 additional deep containers filled with water, and then collected in a 20-liter screened container. The pupae were transferred to a chamber and dried by a slow stream of air. Severe agitation of the

pupae during flotation or air drying was avoided to prevent pupal mortality which should not exceed 5%.

ADULTS

When the pupae had dried, some were used to replenish the fly colony. The remainder were either used as food for pupal parasites, or held for special testing as adults. The fly colony was maintained by placing the pupae in a holding cage (45 x 35 x 35 cm) constructed of aluminum framing covered with plastic mesh window screening. A sleeve of tubular orthopedic stockinette was placed at one end for easy access to the cage. The cage easily held the 5,000 flies with little mortality over an extended period of time. The adult diet consisted of a dry nutrient mixture consisting of 2 parts granulated sugar, 2 parts non-fat dry milk and 1 part powdered egg yolk. Water in a 1-liter waxed container was also placed in the cage. The surface of the water was partially covered with styrofoam chips, to provide greater access to the water by the flies and to prevent drowning. The adult rearing room was maintained at 25.5°C and >60% relative humidity. Using this technique it is possible to consistently rear 1,000,000 house fly pupae per day per week.

REFERENCES

Anonymous.
1958. Aerosol test method for flying insects. In Soap and Chemical Specialties Blue Book and Catalog. 227-228.

Morgan, P. B.
1980. Mass culturing three species of microhymenopteran pupal parasites, Spalangia endius Walker, Muscidifurax raptor Girault and Sanders, and Pachycrepoideus vindemiae (Rondani) (Hymenoptera: Pteromalidae). VIII Renunion Nacional de Control Biologico. Tecomon, Colima, Mexico. April 22-25, 1980.

Morgan, P. B.; LaBrecque, G. C.; and Patterson, R. S.
1978. Mass culturing the microhymenopteran parasite, Spalangia endius Walker. J. Med. Entomol. 14: 671-673.

Morgan, P. B., and Patterson, R. S.
1978. Culturing microhymenopteran pupal parasitoids of muscoid flies. pp. 32-33. In Facilities for Insect Research and Production. USDA Tech. Bull. 1576.

COMMERCIAL PRODUCTION AND USE OF PREDATORS AND PARASITES FOR FLY CONTROL PROGRAMS

E. J. Dietrick

Rincon-Vitova Insectaries, Inc.

In the late 1960's, Rincon-Vitova Insectaries, Inc. began rearing insects that parasitize larvae and pupae of noxious flies associated with filth habitats. We wanted to use these parasites to help reestablish them on poultry ranches which were essentially void of the natural enemies of flies so that only a minimal amount of pesticide applications would be needed for the control of these noxious flies. Although scores of predatory and scavenger species existed in the untreated environments, the parasitic forms were initially favored because of their ease of mass production and their abilities to seek out the later developmental stages of flies, thus resulting in a direct reduction of adult fly emergence.

The commercial insectaries which rear parasites and predators began when the chemical pesticides that were being used no longer controlled fly populations (Dietrick in press). On some poultry ranches, the producers simply parked their spray rigs, because the flies had become so resistant to the registered pesticides that even repeated applications failed to give any long-term control. Overdependence on pesticides had brought on "genetic resistance" in fly populations and repeated usage of one and, then, another chemical resulted in "cross resistance" so the flies could no longer be killed by many classes of pesticides.

Cooperative efforts by University of California researchers and vector entomologists of various county health departments led to the development of alternative cultural controls. This required the frequent removal of the manure from the ranches to nearby solar drying pads or storage areas or directly to agricultural operations for fertilization. Some large ranchers set up elaborate gas-fed dryers to sterilize these waste products, which in turn were sold as cattle feed or pelletized fertilizers. All of these alternative methods were energy- and labor-intensive. While these methods may have alleviated

the fly problem on the ranches, often it was exported into the communities where the manure was being disposed.

At this time scientists, such as Dr. E. F. Legner, Division of Biological Control, University of California, Riverside, were conducting research on biological control of filth flies. Following the basic research of these scientists, our company began offering a pest control advice (PCA) service, integrating all suitable control procedures and making maximum usage of the natural enemy complex. The remainder of this discussion will describe the development of our integrated pest management (IPM) program on poultry farms in southern California and some of the difficulties faced by commercial insectaries in culturing and marketing beneficial insects.

DEVELOPMENT OF IPM OF FLIES USING PARASITES

For the purpose of this discussion, biological control will mean the use of mass-cultured and/or field-harvested predators and parasites (parasitoids) and various naturally occurring microorganisms that cause epizootics in arthropod populations, which can often be harvested and stored for future use. I exclude from my concept of biological control such expanded meanings as the use of third generation pesticides, so-called behavior-modifying chemicals, and those microbial pesticides that have been registered. The natural enemy complex refers to the full range of organisms: predators, parasites, minor pests, decomposers, competitors, and antagonists that are recognizable in biologically balanced habitats, i.e. the untreated commercially clean ranch that produces marketable products.

Certainly the most important resources to consider in the development of any IPM programs are the cultural practices that create favorable habitats for biological control on the farm. Frequent sampling and assessment of natural enemy complexes on ranches where flies are under biological suppression gave us insight into what the interferences were on ranches where flies were out of control (DeBach et al. 1949). We soon found that on those ranches where the development of resistance was severe and the farmers had quit spraying, the natural enemy complex began to reappear naturally. It was apparent that we had to change the way we were managing the manure and begin using pesticides selectively to kill adult flies rather than applying the pesticides directly to the manure attempting to kill the larvae.

Harvesting techniques were developed to obtain complexes of predators from fly breeding "wet" spots in the manure. Using Berlese-type separators we were able to collect the histerid, staphlinid, and carabid beetles in sufficient number for release in areas where they previously had been destroyed. The housefly, Musca domestica L. was mass-reared to provide host material for mass production of several species of parasites

obtained from the University of California, Department of Biological Control Insectary. We chose to rear exotic strains of species that were observed to be important natural enemies on untreated ranches. The material used was the best stock available, based on the research of Dr. Legner. It seemed logical to expect that using biological strains of exotic species from other habitats would help to broaden the genetic capability of the native species. Also, there might be some hybrid vigor develop from making releases of this kind. Often, several months were required to reestablish the complete natural enemy complex. In part, this was the result of the long life cycles of some of the beetles.

Mass-releases of parasites and predators is still productive as an aid to IPM on new and previously chemically treated farms, even though immigrating adults often enter the habitat quickly once the manure pad is initiated and pesticide interference terminated. To exploit biological control, one must tolerate immature flies in the manure habitat in order to provide a source of food for the natural enemies. Periodic colonizations of additional predatory and parasitic species can help the natural enemy complex be more effective. The high reproductive potential of the filth flies necessitates the augmentation and encouragement of the entire complex of natural enemies in order to reduce the numbers of flies to tolerable levels.

Once the natural enemy complex has been established, it must be retained in the habitat. This is achieved by removing the manure only from alternate rows and by leaving a pad at least 8 inches deep where the manure is removed. In this way, not all of the predators and parasites are removed and a favorable habitat is maintained for them. The pad remaining also aids in the rapid drying of fresh droppings.

The role of natural enemies in fly control is best seen in areas of low fly population density. This usually occurs following biological suppression by the combined attack of predators and parasites. When one follows the ecology of a single "wet" spot, it is apparent that the predators attack the fly eggs and small maggots. If the predators are resident in the manure in the immediate vicinity, the eggs and small larvae are destroyed in great numbers. Often the predators are so numerous that very few maggots survive to become puparia. If the predators are less common, greater numbers of flies escape attack. The numerous small predators eat many eggs and small maggots while the larger species of predators attack the larger maggots. Only one imported speceis, Tachinaephagus zealandicus Ashmead, attacks the large maggots, ovipositing in them prior to pupation. All other known parasites oviposit in or on fly puparia. Spalangia endius Walker prefers fresh puparia, while Muscidifurax raptor Gerault and Sanders can be grown on dead pupae. The pupae is preserved by freezing.

Parasites attack other puparia in addition to those in which they oviposit, so their effectiveness extends beyond the number of parasitized puparia. Such host-feeding and mutilation enables the adult parasites to live longer and lay many more eggs when fly breeding is high. Parasites are extremely density-dependent, but their long life cycle makes them no match for their fly hosts. Warmer weather in the spring speeds the development of flies along with increasing the activity level of the predators and parasites, but the population race is clearly won by flies.

It has been observed that those ranches where natural enemies are present in the manure have biological control of flies sooner than ranches where pesticides have previously been used. At these farms where there were good natural enemy complexes, including all the important predators and parasites plus any that are colonized from insectary sources, samples reveal that predators can destroy up to 80% of fly eggs and small larvae, while the parasites can add mortality of 50 to 80% of the 20% surviving puparia. The total mortality can be as much as 95% control of the immature flies. Often only 5% survive to the adult stage and this population of adult flies can easily be managed using selective baits and traps.

Not all presticides have adverse effects, and certain selective usage of conventional pesticides are less disruptive to existing biological controls. Bait stations using diluted, feed-grade molasses (1 qt. to 3 qt. water) give excellent results (Rooney and McKeen 1974). Fiberglass insect screen is cut and stapled to form a bag which is then stapled to a wood lath so that the bag is supported in an upright position. The diluted molasses is placed in a container and the screen bag is fastened around it to keep all flies from contaminating the fermenting brew, and causing putrefaction. (Putrefaction by bacteria limits the life of the attractiveness of the bait.) Finally, the bag is wetted and sprinkled with a sugar bait containing an insecticide. Adult flies seek the water and the brewing yeasts, but are only allowed contact with the poison sugar biats. These traps can be activated and placed where most adult flies are congregating. The traps are effective for up to 6 weeks, as long as the water level is maintained. The idea is to locate bait stations in places attractive to adult flies but not to cover the ranch with them. Such bait stations are useful whenever adult flies are present in large numbers. The number of traps should be increased and other selective pesticide use implemented to kill only adult flies.

The mass inundation of any one species of parasite on ranches where other natural enemy resources have been eliminated will certainly help reduce flies, but results are more cost effective when the parasites are "seeded in" by innoculative releases in an IPM program. It is my opinion that using mass-inundation of any one parasite species where flies are out of

control due to natural enemy interference from pesticides is rather unproductive. Releasing parasites is no substitute for using chemicals in these situations. Biological control results from long-term suppression, not quick kills. In addition, variable weather and seasonal changes in the environment will always cause fluctuations in fly populations. Well-managed ranches undergo fewer fly upsets compared with operations where management is difficult or not understood. The economic position of a particular ranch also restricts the efficacy of a management effort. Whatever is done must be cost effective and within the limited budget of the ranch. It is cost effective to teach the IPM techniques along with procedures for releasing the parasites. Periodic colonizations into the fly breeding spots help biological control of flies by natural enemies (Legner and Dietrick 1972). Whatever is done must be done quickly and selectively to suppress pestiferous adult populations of flies with minimal interference to biological control that takes place in the manure. The parasites received from the insectary are best used by placing them in manure in the spots where fly breeding is known to be present. The operator of the ranch knows this better than anyone, and this activity can be part of day-to-day ranch operations. Having ranch personnel do this eliminates the danger of the entomologist carrying diseases from ranch to ranch.

Not all flies are of agricultural origin. Over half of the flies have been shown to come from the neighborhood garbage cans, compost heaps, grass and other prunings, cat and dog droppings, and other backyard animal husbandry. Migration of adult flies onto the poultry ranches gives the agriculturist problems that he must respond to. Bait stations surrounding the ranch or at least in the path of the immigration of adult flies helps control them. Such a ranch, that is located in an urban environment, can be both a trap for adult flies and an insectary that exports natural enemies throughout the neighborhood, wherever fly breeding is taking place. The resulting control may not be as effective as was possible before pesticides began to fail, but it is far more satisfactory than trying to manage the hords of flies that leave a ranch when repeated poisoning is no longer effective.

Like most IPM programs that our company manages, this one developed out of research. Scientists working at university or federal research laboratories find or develop useful biological controls. Our company, working as consultants directly on the farms, integrates those compatible ideas, and often the newly acquired work animals (predators and parasites), blending the techniques into practical programs for our customers. In the case of biological control of flies associated with solid animal waste accumulations, the principal research for our program came from Dr. Legner. His investigations were partly supported by grants from the National Institutes of Health and the U.S. Public Health Service. Our company helped

develop this research for our customers who support pest management using natural enemies.

County health personnel (particularly in San Bernardino County, California) and University of California extension specialists and researchers worked together with PCA's (pest control advisors) from our company and with minimal funding from our growers to establish the scientific basis for programs that grew out of common sense observations (Dietrick 1972). Making maximum use of predators and parasites, we have continued to expand our IPM program to other habitats (dairies, cattle feed lots, etc.). Unfortunately, we have not been able to put together the cooperative research effort in all situations.

In the meantime, our repeated observations and recordings of recurring natural happenings continue on the ranches where we are practicing IPM using parasites. In applying the techniques, the PCA's can repeatedly assess the ranches in their care. This constitutes a continuous experiment in applied ecology not duplicated at the research level. This experiment is carried out in many different field situations, throughout the season, and often over many years on the same farms. All levels of pest density or problems are assessed, including clean fields with no problems. What better proof is there of success of IPM than the practical experience of profitable production of quality products and the favorable testimonials of our clients in support of our efforts?

CULTURING AND MARKETING FLY PARASITES

Like farming, the growing of many different species of insects for market has great complexity peculiar to each species. There may be several equally effective ways to mass-culture any one of them. When raising parasites of filth flies, one must choose a species of fly to serve as a surrogate host for the parasites in the insectary. We chose the housefly, Musca domestica L., because it is cosmopolitan in distribution and one of the principal target pests.

The method of mass production was developed by our insectary manager at the time, though it has been modified many times (M.E. Badgley unpublished). The flies are fed on a mixture made from calf replacer dried milk and wheat bran mixed with rice hulls. The mature maggots are driven from the media before pupation by excessively wetting it. This procedure takes advantage of the tendency of fly larvae to crawl to a dry place in which to pupate. The larvae move readily to trays where fans dry them and they begin to form puparia. With this method we have a source of both mature maggots and puparia of all ages for use in the parasite production.

Tachinaephagus zealandicus requires larvae for oviposition (sting). This exotic, multiple parasite lays its eggs in maggots as they migrate from wet spots to dry areas for pupation. We are still interested in this species, since it is the only parasite that attacks larvae. Cool, moist conditions must be maintained to satisfactorily rear this species in the insectary.

Spalangia endius parasitizes fly puparia by puncturing the outer wall and ovipositing on the pupal body. The egg hatches, and the parasite larva attaches itself to the pupa. It is a solitary parasite that does well at higher temperatures over a wide range of geographical areas.

Muscidifurax raptor is similar to Spalangia, but it seems to prefer our coastal climates in California and is deterred by hot conditions in mid-summer. We colonize these during spring and fall and in cooler climates.

Other species have been reared, including Pachycrepoideus vindemiae (Rondani) and Muscidifurax zaraptor Kogan and Legner and M. raptorellus K. and L. Several species of histerid beetles have been propagated, but the economics of feeding predators that have life cycles of several months and eat large amounts of fly eggs has questionable cost effectiveness, except where classical biological control (i.e. the introduction of new, exotic species) is concerned. All possible efforts should be made to import the several species of histerid beetles that are known to help biological control of flies throughout other continents. Our insectary supports these efforts at importation in every way possible; because when these exotic forms become established, they are then useful to our customers in the IPM programs that we offer.

Rising energy costs are affecting both the insectary production and the delivery of IPM services. Rising automobile operating costs now limit our travel to and from the ranches. We are relying more on the postal and various other delivery systems to move the parasites to the customers. Our service is now more educational and trouble shooting, with more time spent training ranch personnel to implement IPM techniques as needed on their farms. While our emphasis on service has decreased, our business has become more oriented toward our products and over-the-counter sales to independent IPM advisors and other professionals who must travel to the ranches for other purposes, e.g. veterinarians, nutritionists, etc. In addition, we have attempted to make our mass-production more cost effective.

Such indirect sales lessen our control over the products, but the programs have to be cost effective if they are to provide the successful alternatives to failing existing programs. Many of our customers are teams of PCA's who service a well

defined area with intensive programs to deliver a wide range of IPM products and services. Some may operate their own insectaries. In this way, maximum use of habitat management techniques that are capable of trapping pests and increasing the populations of predators and parasites can then be exploited. Agricultural pests are everybody's business in an agro-ecosystem, and the destruction of natural enemy complexes in nearby fields affects all situations, particularly those farmers who are practicing biological control. Drift of poisons directly onto the ranches as well as the disposal of other agricultural wastes can cause fly problems on otherwise well managed ranches.

Probably the single most prevalent problem facing the commercial insectary is the low visability of biological controls. It would help greatly if extension personnel would provide the farmers with equally positive information regarding description and behavior of major natural enemies as they presently now concentrate on pests. There are abundant guides for using pesticides to kill everything, with little regard for the useful predators. There are few bulletins that provide "do-it-yourself" programs that describe the main natural enemies of various pests or any recipes for enhancing their effectiveness in any habitat. Unfortunately, concerning the mass release of beneficial insects, the usual statement from the entomological establishment, including many researchers in biological control, is that "it won't do any harm to use them." Weak statements that call for conservation of natural enemies without identifying the species involved, or without offering advice about what options are available, or what will be the probable consequences of conventional programs that no longer provide adequate control are worthless. Some pest situations are so unresolvable today that the option to do nothing can be better than creating more problems with the cures that are offered.

Like most businesses marketing products with no shelf life, the insectary must have a steady market to survive. Once the insects are grown, they must go to market or the quality of the product is lost. Other pest controls can be kept in readiness for problems to develop. The insectary functions best when prearrangements are made for an orderly delivery of products as they emerge from the production cages, much like milk or eggs. This is the reason for following the programs of periodic colonizations during the fly breeding season, rather than trying to time a perfect single mass-release. The insectary can best offer a steady flow of products to the grower to be placed wherever fly breeding is taking place during that particular week. Many small releases have a better chance of successful augmentation of the natural enemy complex than trying to put them all out at once. Pricing of the product has to cover the loss of all parasites not sold, as well as the year-round maintenance of cultures in the off-season.

We have had more than ten years of successful IPM using parasites on poultry farms. Acceptance by county health department vector entomologists and the farm owners has become widespread. Even if the program is not an immediate success in all cases, suppression of fly problems in the long run can be achieved when natural enemies are restored to the ranches and the predators and parasites increase in numbers to change the balance in favor of biological control. Selective use of pesticides and baits can shorten the time required arrive at a favorable balance. Every adult fly killed is several hundred fly eggs that need not be devoured by predators and parasites.

Success comes suddenly when the number of fly adults emerging from the breeding spots begins to decline. Fly sticky tapes can record this dramatically. The number of complaints drop, cleanout costs are visably reduced with fewer wet spots where physical removal of maggot infested manures that must be disposed of occur, and fewer explosions of adults occur. Pesticides and application costs are considerably reduced. Probably the biggest benefit occurs for the ranch personnel, who no longer are repeatedly exposed to residues of poisons. The owner can also rest assured with the knowledge that his products will not be rejected for pesticide residues. Even most veterinarians recognize the reduced stress placed on the animals by reducing their exposure to broad spectrum pesticides.

REFERENCES

DeBach, P.; Dietrick, E. J.; and Fleschner, C. A.
1949. A new technique for evaluation the efficiency of entomophagus insects in the field. J. Econ. Entomol. 42: 546.

Dietrick, E. J.
1961. An improved backpack motor fan for suction sampling of insect populations. J. Econ. Entomol. 54: 394-395.
1972. Private enterprise pest management based on biological controls. Tall Timbers Conf. on Ecol. Animal Control by Habitat Manage., Tallahassee, Fl. 4: 7-20.
Commercial production of entomophagus insects and their successful use in agriculture. Beltsville Symp. Biol. Control Crop Prod. V: (in press)

Legner, E. F., and Dietrick, E. J.
1972. Inundation with parasitic insects to control filth breeding flies in California. Proc. Calif. Mosq. Control Assoc. 40: 129-130.

Rooney, W. F., and McKeen, W. D.
1974. Molasses looks good for fly control. Agric. Ext. Serv. Univ. Calif., San Bernardino Co.

Schlinger, E. I., and Dietrick, E. J.
1960. Biological control of insect pests aided by strip farming alfalfa in experimental program. Calif. Agric. 14: 8-9.

THE USE OF PARASITES AND PREDATORS IN COMMERCIAL INTEGRATED PEST CONTROL METHOD FLY CONTROL PROGRAMS

Richard C. Frey

Arizona Biological Control, Incorporated

INTRODUCTION

For the past three years Arizona Biological Control, Incorporated has been in the business of biological fly control. The need for alternative methods of pest control coupled with a keen desire to manifest my skills as a population biologist prcvided the perfect environment for development of business strictly dealing with biological control.

Arizona Biological Control, Inc. (ARBICO) currently treats more than 300 customers in the states of Arizona, California, New Mexico, Texas, Nebraska, Colorado, Oklahoma, Pennsylvania, Illinois, Wisconsin, Utah, and the countries of Mexico and Panama. These customers consist primarily of feedlots, dairies, poultry farms, hog farms, and horse operations. ARBICO recently branched out into the areas of human sewage treatment plants and landfills. Parasites can essentially be used in any environment where organic decomposing material is found.

The following represents seven case studies which have been successfully treated and continue to be treated using an integrated pest management program.

CASE STUDY 1--A CAGED LAYER FACILITY IN HERMOSILLO ,MEXICO-- HARBORING 160,000 LAYERS

This commercial facility suffered from two major problems: a lack of waste management and disposal problems. The facility was brought to our attention because it is located within the city limits and many of the nearby dwellers had been complaining about the flies. Upon arrival to the site, we confirmed that it was one of the worst situations we had ever encountered but felt we would be able to assist in reducing the problem.

The first object was to improve the manure management by re-

moving the dry waste from under the cages and leaving a dry pad of ten to fifteen centimeters of manure before the hot summer months arrived. Secondly, the water system was corrected by fixing leaky troughs, cups, and implementing a soft water system to prevent corrosion.

Thirdly, a weekly introduction of fly parasites were introduced into the environment. Within approximately 6-8 weeks the problem had reduced by 60-80%. The neighbors and public health officials were impressed with the results and have continued to the present to recommend this program of integrated pest management to other commercial as well as government firms raising caged layers, dairies, feedlots, and swine farms throughout Mexico.

CASE STUDY 2--A DAIRY IN ARIZONA WITH 1500 HEAD

The main problem with this dairy was centered around the lack of manure management. Although the dairyman was assuming he was disposing properly of the manure, an important "hot spot" was overlooked time and time again.

There were 50 to 100 calves that were confined in pens which were not being cleaned out on a regular basis. Hay was being added regularly to insure the calves comfort, while at the same time providing an excellent breeding area for the flies.

Upon visual notification of this "hot spot" the dairyman soon began to clean the pens more carefully. Weekly releases of fly parasites coupled with occasional chemical spraying reduced the problem by 65-75%. The dairyman felt the program had increased milk production.

CASE STUDY 3--A FEEDLOT IN ARIZONA WITH BETWEEN 10,000 TO 19,000 HEAD

This feedlot was called upon during the month of June. The severity of the fly problem was evidenced by the fact that the cattle chose to stand in the 105 degree sun in lieu of seeking the comfort of the shade awnings due to the heavy infestation of flies located there. The constant switching of tails as well as twitching their bodies also showed that even in the blistering sun the flies were still a nuisance.

Upon making a thorough survey of the feedlot, we recommended that a corrective program of manure management as well as weekly releases of fly parasites be implemented.

This feedlot has now been using an integrated approach for the last three years. In the past it had been necessary to spray once every other day; now the spraying occurs monthly. Due to the reduction in spraying it was necessary to employ one

extra person just to do the spraying. They felt that the IPM method saved them between 30-40% of their overall operating cost.

CASE STUDY 4--USE OF IPM AT THE UNIVERSITY OF ARIZONA

It was and still is felt that one of the most important factors when entering a new field is to communicate closely with University as well as Cooperative Extension officials. The University offered the opportunity to execute experiments in three dynamically diverse areas: caged layers, a dairy, and a feedlot.

We were able to sample pupae and discover the level of parasitation before commencing the program and after the start of the program much was learned about "hot spots" and new techniques of distribution were employed. In every instance a control group was set aside to compare results.

The University agreed to begin the program for the first year and not continue the second year to check the results of not using IPM. This year they looked forward to the use of the IPM method as the previous year the flies had been exceptionally bad. They felt the use of IPM was instrumental in the overall success of the previous year's program.

CASE STUDY 5--A MEATPACKING FACILITY IN ARIZONA

This slaughterhouse has the reputation of being one of the cleanest in the state. It harbors at any single time between 200-500 head in close confinement pens.

The use of a weekly dose of fly parasites during a period of three years has caused as the employees stated "a drastic improvement, resulting in dollar savings with respect to chemical usage, less meat loss".

Slaughterhouses such as these depend greatly on the IPM method. If a health inspector detects the presence of flies within the confines of the plant, it can be justification for closing the operation resulting, in many cases, in tremendous financial loss.

CASE STUDY 6--A RARE BREED CANINE FACILITY

IPM has been used in the case where canines are confined in pens for long periods of time. Education on the techniques of manure management as well as a weekly dose of fly parasites has kept this customer virtually "fly free".

CASE STUDY 7--A SWINE FARM IN NORTHERN ARIZONA

The most important reason to use an IPM approach on these types of facilities is to insure that the swine can be kept disease free. The fly presents a menace to swine breeders with its dynamic capability to spread disease.

The particular situation was treated by use of the fly parasites around the circumference of a nearby lagoon which was used to store the manure which would be used ultimately as fertilizer. A weekly release of insects in the aforementioned area and in other "hot spots" gave the customer the control he was looking for. He will continue on the program this year.

CONCLUSION

It is clear to see that the IPM method has merit. However, as a commercial producer of these insects and as a firm believer in IPM the most important issue to bring out is that in order for this type of program to be a successful one the users of this program must be educated.

The potential customer must be taken out into the field and shown how flies breed, by personally demonstrating where the breeding sites are located, picking up pupae, locating "hot spots". In addition, a careful survey of the manure situation must be carefully made. If there are problem areas with the management and disposal of the manure, the potential customer must understand the program will fail if a totally integrated approach is not used. Occasional sprays or non-toxic traps should also be encouraged to knock down the adult flies if needed.

Once the decision to use an integrated program has been made, the customer is taught how to implement the program according to their particular needs.

In order to reduce the human error, our representative will personally instruct the customer or their employee where to release insects, when to release the insects, how often to release the insects, and how to look for results. A manure management consultation will also take place the first time an order is delivered.

The representative will then call on the customer on a monthly or bi-monthly basis depending on the particular needs they might have. Unfortunately, human error is responsible in the vast majority of cases where the IPM method apparently fails. Therefore to insure a successful program, the regular visitations are crucial as well as a quarterly questionnaire which aids to determine the success of the program; if the customer is receiving an ample supply of insects, if the

insects are arriving regularly, if meteorlogical conditions warrant an augmented or depleted dosage, if the amount of animals has increased or decreased.

Quality control is another important factor that will insure the success of IPM. If the parasitation is not between 80-100% the insects will potentially be unable to do the job they were intended for. Therefore, it is and should be the policy of all commercial insect growers that insects be of the highest quality.

Cost is another factor not to be left unconsidered. The program must also be cost effective and affordable. The aim of commercial growers should be a program of economical fly control.

In concluding it is my belief that there is nothing which works better and is more cost effective than the IPM method. The success of ARBICO'S use of IPM has depended on three essential factors: maintaining contact on a regular basis with customers, using quality affordable insects, and educating as to the techniques of good manure management. I feel if these basic requirements persist the commercial growers of insects will continue to maintain a good reputation in the field.

THE COMMERCIAL INSECTARY IN TODAY'S INTEGRATED FLY CONTROL PROGRAMS

R. Pewitt

Agricultural Insect Management

Since 1889 the agriculture industry has been using some form of beneficial insect in programs to control pests. Two of the major problems facing the users of these natural enemies has been the availability of the beneficials and the cost of large numbers, once they were found. Today, the successful use of fly parasites to control filth breeding flies has brought into existence a number of commercial facilities producing several species of these tiny natural enemies. The competitive environment of the free enterprise system has brought about higher quality insects and lower prices, making them available to virtually everyone who must control pest flies.

First, it must be pointed out that the use of fly parasites is only one component of an overall Integrated Pest Management Program (IPM). The program consists of a few common sense steps to eliminate fly breeding sites to make the poultry farm, dairy, hog unit, feedlot, etc. less of an ideal environment for flies. Some of these steps include:

(1) Good sanitation is primary to an effective integrated fly control program. Water leaks should be fixed promptly, as well as, manure composted or spread to dry. Spills of feed, milk and broken eggs should be dealt with appropriately.

(2) The overuse and incorrect application of many chemicals has not only rendered those chemicals useless but created a "super" pest. As a result, many larvacides only provide a moist environment for pest "super" flies to breed in. Therefore, restraining the use of chemical pesticides will enhance the biological control program and prolong the effectiveness of the pesticides.

(3) Seeding the manure and other non-manure fly breeding areas with large numbers of natural enemies creates a hostile environment to the remaining fly population.

Now the customer has found the fly parasites are available to him. He has decided an integrated biological program will be best for his facility. However, there are several choices involved in the application of the insects. There is the pest control operator (PCO). This choice frees the customer from decisions about where to put the insects, how many to release, and what species is best for his facility. The PCO also has the equipment to use residual or knockdown sprays to deal with adult flies that may migrate from other areas and the awareness not to damage the parasite and other beneficial insect populations. Another choice is an IPM consultant. This may be an entomologist or a pest control adviser. In addition to the fly parasites, the customer is paying for advice on sanitation and the use of fly parasites. Generally, the adviser or consultant will initially survey the facility to be treated and make suggestions on methods of eliminating breeding areas and on the application of the fly parasites. In this case, the customer will release the insects himself. He usually owns his own chemical application equipment to use if necessary. The third choice is dealing directly with an insectary that raises the fly parasites. Most insectaries offer general advice on sanitation and complete instructions on the application of the natural enemies. The main advantage in dealing directly with the insect producer is economic. The cost of the parasites is usually much less because there is no middleman and the advice and instructions are free. This is particularly advantageous to poultrymen where people control can be a major problem. This choice puts the most responsibility for fly control on the farmer.

In all of these options there is one basic necessity. The insects must be a constantly, high-quality product. It is impossible for an adviser, consultant or an insectary to make decisions concerning the species and quantity of insects to apply when the supply is inconsistant and the viability is poor. In the earlier days of the fly parasite industry, several organizations sold a good idea with an inferior product and the industry took a giant step backwards. Even USDA and university research results on application, numbers required, and species effectiveness can be misleading, if the quality of the fly parasites reared or purchased is unknown.

To the commercial insectary, all the choices discussed--the pest control adviser, the professional consultant, the pest control operator, and the grower--are potential customers. They all must be properly educated about what quality fly parasites are, how to apply them, how many insects will work in various situations, and which species are best for each

situation and geographical location. Cooperation with researchers to more accurately determine these various points and to continually upgrade quality is essential. Because of input from all of these avenues, the commerical insectary can be an excellent pool of information concerning sanitation and manure management techniques, as well as, other points of an integrated fly control program.

AFTERWORD

It is clear from this meeting that more research must be done on the practical cost-efficient use of parasites and predators in the field and under various conditions (most work has been done on poultry farms). The commercial people were criticized because many times their product was not as advertised and was often a species that is noneffective in the field. All research indications are that *Nosomia vitropennis* is not an effective parasite in the field, but is often sold because it can outcompete the other species in the colonies. The main criticism of this workshop was the lack of time for discussions. Most participants seemed to enjoy the one-night discussion session when many topics were discussed very openly and frankly without malice. Any future workshop should allow more time for this type activity.

I would like to see another workshop held in another three to four years. I do not feel that an annual meeting would be useful as the research is slow and it could become repetitious. If you, the participants of this workshop, feel it was useful and would like another, let us know and we perhaps can initiate plans for another workshop in the future.

R. S. Patterson
Chairman, Organizing Committee

THE AUTHORS

Axtell, Richard C., professor of entomology, North Carolina State University, Raleigh, NC 27650

Butler, J. F., professor of entomology, University of Florida, Gainesville, FL 32611

Campbell, John B., professor of entomology, University of Nebraska, North Platte, NE 69101

Dietrick, E. J., president, Rincon-Vitova Insectaries, Inc., Riverside, CA 92506

Drea, J. J., Jr., NRPL, Biological Control of Pests, Science and Education Administration, U.S. Department of Agriculture, Beltsville, MD 20705

Escher, R. L., entomologist, University of Florida, Vero Beach, FL 32960

Frey, Richard C., president, Arizona Biological Control, Inc., Tucson, AZ 85704

Gersabeck, E. F., graduate student, Department of Entomology, Michigan State University, East Lansing, MI 48824

Harris, R. L., research entomologist, Veterinary Toxicology and Entomology Laboratory, Science and Education Administration, U.S. Department of Agriculture, College Station, TX 77840

Hogsette, J. A., research entomologist, Insects Affecting Man and Animals Research Laboratory, Science and Education Administration, U.S. Department of Agriculture, Panama City, FL 32401

Hoyer, H., entomologist, European Parasite Laboratory, Science and Education Administration, U.S. Department of Agriculture, c/o American Embassy--Agriculture, APO New York 09777

Kennedy, M. K., professor of entomology, Michigan State University, East Lansing, MI 48824

King, E. G., director, Bioenvironmental Insect Control Laboratory, Science and Education Administration, U.S. Department of Agriculture, Stoneville, MS 38776

Koehler, Philip G., professor of entomology, University of Florida, Gainesville, FL 32611

Legner, E. F., professor of entomology, University of California, Riverside, CA 92521

Loomis, Edmond C., professor of entomology, University of California, Davis, CA 95616

Merritt, R. W., professor of entomology, Michigan State University, East Lansing, MI 48824

Miller, R. W., research entomologist, Livestock Insects Laboratory, Science and Education Administration, U.S. Department of Agriculture, Beltsville, MD 20705

Morgan, Philip B., research entomologist, Insects Affecting Man and Animals Research Laboratory, Science and Education Administration, U.S. Department of Agriculture, Gainesville, FL 32604

Nolan, Maxcy P., Jr., professor of entomology, University of Georgia, Athens, GA 30602

Patterson, R. S., research entomologist, Insects Affecting Man and Animals Research Laboratory, Science and Education Administration, U.S. Department of Agriculture, Gainesville, FL 32604

Petersen, J. J., research entomologist, Livestock Insects Research Unit, Science and Education Administration, U.S. Department of Agriculture, Lincoln, NE 68583

Pewitt, R., president, Agricultural Insect Management, Grady, AL 36036

Pickens, Lawrence G., research entomologist, Livestock Insects Laboratory, Science and Education Administration, U.S. Department of Agriculture, Beltsville, MD 20705

Rutz, Donald A., research associate, Department of Entomology, North Carolina State University, Raleigh (present address: Cornell University, Ithaca, NY 14853)

Stage, D. A., graduate student, Department of Entomology, Science and Education Administration, U.S. Department of Agriculture, Lincoln, NE 68583

Thomas, Gustave D., research entomologist, Biological Control of Insects Research Laboratory, Science and Education Administration, U.S. Department of Agriculture, Columbia, MO 65211

Weidhaas, D. E., director, Insects Affecting Man and Animals Research Laboratory, Science and Education Administration, U.S. Department of Agriculture, Gainesville, FL 32604

☆U.S. GOVERNMENT PRINTING OFFICE: 1981—771-039/1C

NATIONAL AGRICULTURAL LIBRARY
1022312100

U.S. DEPARTMENT OF AGRICULTURE
SCIENCE AND EDUCATION ADMINISTRATION
P. O. BOX 53326
NEW ORLEANS, LOUISIANA 70153

OFFICIAL BUSINESS
PENALTY FOR PRIVATE USE, $300

NATIONAL AGRICULTURAL LIBRARY
1022312100

POST
U.
AGRICULTURE
AGR 101

U.S.MAIL